RECIRCULATING AQUACULTURE

$$Q_1 C_2 - Q_1 C_1 = P_x$$

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J.M. Ebeling

Northeastern Regional Aquaculture CENTER

Recirculating Aquaculture 2nd Edition

By

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Recirculating Aquaculture 2nd Edition

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DEDICATION

We would like to co-dedicate this new text to Dr. Wheaton (Fred) and Dr. W.D. Youngs. Fred was James Ebeling's major professor for his PhD at University of Maryland. In fact, many of us in the industry had Fred as our major advisor. Fred was way ahead of his time In 1977 and wrote what became known as the Aquacultural Engineer's Bible, "Aquacultural Engineering", in 1977. This text was the foundation for many teaching programs around the world and remains popular still. Most of the technical information published in this 1977 text remains valid today 30 years later! Fred was a co-author on our earlier text in 2002 "Recirculating Aquaculture Systems" by Timmons, Ebeling, Wheaton, Summerfelt and Vinci (2002 by Cayuga Aqua Ventures, Ithaca, NY).

Fred has chaired over 50 graduate committees. Many of the PhD students have gone on to be the leaders in the aquaculture research and teaching community. Fred has always been a very supportive individual and has continued to mentor us as we progress through our careers.

Dr. Wheaton developed one of the first aquacultural engineering research and extension programs in the U.S. His research has included recirculating systems, seafood processing, automation of oyster shucking, and a variety of other topics related to aquacultural engineering. He has published widely producing over 100 articles and three books. He was one of the founding members of and has served as president of the Aquacultural Engineering Society. Dr. Wheaton recently retired (June 2010) as Director of the USDA Northeastern Regional Aquaculture Center (5 years) and was formerly Chairman and Professor, Department of Biological Resources Engineering, University of Maryland, College Park, Maryland; Dr. Wheaton was a faculty member for 42 years at the University of Maryland.

We all owe a great deal of gratitude to Dr. Wheaton for his career efforts as a developer and supporter of the aquaculture community. For this and many other reasons, we dedicate this book to him.

Our second dedication goes to Dr. William D. Youngs, Professor Emeritus, Department of Natural Resources, Cornell University. Dr. Youngs spent over 30 years at Cornell teaching and mentoring students in fishery science and aquaculture. Dr. Youngs is most recognized for one of the seminal texts in fisheries, "Principles of Fishery Science", that he co-authored with Dr. W. Harry Everhart, a book that many keep in their personal libraries (published 1981, Cornell University Press, p. 349).

For me (Timmons) it was a phone call from Bill in 1984 that was my introduction to the world of aquaculture and, in particular, recirculating aquaculture. The phone call went something like "What size pump do I need to move 1,000 gpm of water against 10 feet of head?" I responded with my own question, "Why do you want to know that?" Bill took me under his wing and we spent many hours together building the first recirculating aquaculture system at the Cornell Dairy Research farm. Bill was also the first General Manager of my commercial aquaculture venture, Fingerlakes Aquaculture (see Chapter 17).

Bill has always been a constant inspiration to me. I truly value Bill's advice on a wide variety of subjects, but I consider him unequaled in experience and scientific knowledge on fisheries management as applied to recirculating aquaculture. I also have the honor of being the only person that Bill ever took fishing and failed to even have a fish strike! We hope to eliminate that distinction soon.

Thanks Bill!

July 2010

FOREWORD

Aquaculture has a long history with its origins dating back to at least 475 BC in China (Milne, 1973). Trout culture started in Germany in 1741 (Leitritz and Lewis, 1980) but it wasn't until the 1880's that trout culture came to the U.S. This was the first U.S. aquaculture effort. However, aquaculture was not of much importance until the late 1940s when it was discovered that aquaculture methods could be used to raise fish for planting in natural waters as a means to supplement natural spawning. At this time the U.S. fish and Wildlife Service began growing trout (*Oncorhynchus mykiss)*, bluegill (*Lepomis macrochirus)*, largemouth bass (*Micropterus salmoides)*, and other species for planting. From its U. S. initiation in the 1880s the U.S. the trout industry grew slowly until the late 1940s or early 1950s when it began to expand more rapidly. In the 1960s the U.S. catfish (*Ictalurus punctatus*) industry got its start and began to grow rapidly. Although the catfish industry had its ups and downs over the years, it is an excellent success story. The catfish industry started from essentially no commercial production in 1960 to over 600 million pounds of production in 2000. With the rise of the trout and catfish industries in the U.S. there has been experimentation and now commercial production of many other species of fish including but not limited to striped bass (*Morone saxatilis*), salmon (several species), yellow perch (*Perca flavescens*), tilapia (several species), blue gill, small mouth bass (*Micropterus dolomieu*), several species of bait fish, goldfish (*Carassius auratus)*, koi, red fish, sturgeon (several species), a wide variety of tropical fish, and a variety of other fish species. Tremendous progress has been made in fish culture in development of production systems, nutrition, genetics, engineering, disease control, physiology, basic understanding of fish biology and other areas. However, much remains to be done in all of these areas.

Shellfish aquaculture has been practiced in the U.S. since about the 1850's, but serious culture really became more popular only in the last 30 years. Species such as blue mussels (*Mytilus edulis*), various species of oysters and clams, shrimp, lobsters, crabs, and others have been raised in aquaculture settings. Most shellfish aquaculture in the U.S. uses leased bottom systems, raft and rack culture, up-wellers, and/or some floating systems using either lines or cages. U.S. shellfish culture is almost exclusively done in salt or brackish water while, except for salmon, most fish culture is in fresh or brackish water. However, salt water fish culture is rapidly increasing. Development of shellfish culture is lagging behind fish culture in areas of nutrition, engineering, disease control, genetics, basic

biology and other areas.

Aquaculture has been the fastest growing segment of U.S. agriculture for more than 15 years and is projected to remain that way for the foreseeable future. This rapid growth rate is driven by several factors including: 1) many fisheries have reached their sustainable yield, 2) food safety concerns of consumers, and 3) consumer demand for high quality, safe aquatic products that are low in fat and high in protein.

The consumer trend that sees more meals eaten away from the home also contributes to aquaculture production as most seafood is eaten in restaurants and other eating places. These businesses need a reliable supply that can provide products on a regular basis year around, something a natural fishery can rarely provide.

Although the future of aquaculture is bright in the U.S. there also are risks. Regulation of water supplies (both quantity and quality), waste discharges, and health regulations are becoming more and more onerous and costly to the industry. Competition for coastal sites, the public's concerns about environmental factors ranging from pollution to concerns about the visual "pollution" of aquaculture facilities in front of vacation homes on the shore, and the recreational use of waters that are also suitable for aquaculture are only a few of the clouds on the aquaculture horizon. These and other concerns are encouraging the aquaculture industry to move from open pond and cage culture systems to the more closely controlled recirculating systems.

Typically recirculating (closed) aquatic production systems have higher capital and operating costs than many of the extensive systems such as cage culture in natural waters and raceway and/or pond culture systems. However, when the control provided by recirculating systems and the benefits this environmental control provides in terms of marketing, waste control, product quality, product availability, and other factors are considered-- then recirculating systems become much more attractive. Thus, this text is designed primarily for recirculating systems, which the authors feel will be the systems of choice for most new aquaculture ventures. The information provided in this text does, however, also apply to open, semi-closed, and closed systems.

The objectives of this text are the practical application of aquacultural engineering and how to design, construct, and manage an aquatic production system. It provides the reader with essential information necessary to get started in aquaculture production and it emphasizes practical information rather than in-depth theoretical discussions. It does not provide the reader with information on genetics, basic biology, marketing, and all of the other areas important to development of a successful aquaculture operation. Many of these topics are touched on in the text, but

are presented only in sufficient detail to allow the reader to understand the relationship of each of these aspects to production of fish. There is no attempt to present in-depth discussions of these topics. Rather the object is to provide sufficient information so the reader can: 1) look at a system and make a good judgment as to how well the systems will operate, 2) work with a systems designer to develop an aquatic production system of your own, and 3) know what to look for when shopping for aquacultural production systems.

The authors of this text combined have over 50 years of experience in aquacultural engineering.

Michael B. Timmons, Ph.D. Dr. Timmons received his B.S. in Agricultural Engineering from the Ohio State University, his M.S. in Agricultural Engineering from the University of Hawaii, and his Ph.D. from Cornell University. Dr. Timmons has worked in aquacultural engineering for 25 years as a researcher, teacher, and extension specialist. He has published widely and has served as primary editor on many of the Aquacultural Engineering Society meeting proceedings and for the series of bi-annual meetings sponsored by Virginia Tech on Water Recirculation Systems. He was one of the founders of the Aquacultural Engineering Society and has served in several officer positions including President. Dr. Timmons was a J. Thomas Clark Professorship of Entrepreneurship and Personal Enterprise (1999-2006) at Cornell University where he is still a professor in the Department of Biological and Environmental Engineering. Dr. Timmons has been a principal investor (he put his house on the line!) in the design, construction, and operation of a commercial recirculating tilapia farm (~500 tons per year of production) and thus provides the viewpoint of a commercial aquaculturist in addition to his experience as a researcher and extension specialist.

James M. Ebeling, Ph.D. Dr. Ebeling has a B.S. and M.S. in physics from Albion College in Albion, Michigan and Washington State University in Pullman, Washington, respectively. He has a second M.S. in agricultural engineering from Washington State University and has three years of formal training at the University of California, Davis in aquacultural engineering. He obtained his Ph.D. in

Biological Resources Engineering from the University of Maryland, College Park, Maryland, where he worked on the kinetics of biofilters operating on aquacultural systems. In November 2006, James was selected as a Fulbright Senior Specialists Candidate (Council for International Exchange of Scholars, Washington DC)

Dr. Ebeling has been involved in aquaculture for over 25 years and has cultured over 20 species of fish. He spent three years at the Mariculture Research and Training Center, University of Hawaii as a research coordinator, and one year as project manager for the design and construction of the "Fish Barn" at the North Carolina State University. Dr. Ebeling also spent five years as a research and extension associate at the Piketon Research and Extension Center, Ohio State University, Piketon, Ohio where he was responsible for design, construction, and maintenance of the aquaculture facilities and for maintaining the fish as well as for the Center Aquaculture Extension Program. He spent six years at the Freshwater Institute as an Environmental Research Engineer, working in basic and applied research as well as the application of monitoring and computer control to biological and aquatic ecosystems. Dr. Ebeling is currently employed as a Research Engineer by Aquaculture Systems Technologies, LLC, New Orleans, LA. E-mail: jebeling@Beadfilters.com; phone 504-837-5575.

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We would also like to particularly thank Mr. Joe Hankins, Director and the Conservation Funds Freshwater Institute (Shepherdstown, WV) who supported the development and writing of the earlier book "Recirculating Aquaculture Systems" (published in 2002) and who co-sponsored the Cornell-Freshwater Institute Short Course from 1999, which instigated the writing of the first text (we got tired of printing so many handouts, so we wrote the book instead). The first text was financially supported in part by the Northeastern Regional Aquaculture Center and continues to distribute the book. Also to the NRAES (Natural Resources, Agriculture, and Engineering Services, a Land Grant Outreach program, Ithaca, NY; see www.NRAES.org for a complete list of available publications and resource materials) who permitted the use of some materials previously published in NRAES-49 "Engineering Aspects of Intensive Aquaculture".

We owe a deep debt of gratitude to Dr. Raul Piedrahita (Professor Department of Biological and Agricultural Engineering, University of California, Davis, CA 95616 U.S.A. phone 530-752-2780, E-mail rhpiedrahita@ucdavis.edu) who carefully edited the entire text, chapter by chapter (this was to be able to translate the entire text into Spanish, currently available from www.c-a-v.net), and made many thoughtful suggestions and corrected many mistakes we had over-looked. Thank you Raul!! And, then Mr. Erik Kopache (my recent graduate student) did a final text editing and conversion of previous files into camera ready format for the printer. Thank you Erik!

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CHAPTER 1
INTRODUCTION TO RECIRCULATING AQUACULTURE TECHNOLOGY

1.0 BACKGROUND

Recirculating Aquaculture Systems (RAS) have evolved over the past thirty years through research and development by university and commercial research and demonstration facilities and through continuous refinement of each subsystem process. The two primary authors and the other contributors to this text have been at the forefront of this activity. The focus of our work has been on developing RAS that can produce food fish on an economically competitive basis. But in addition, the content of this book has generally applicable to all forms of aquaculture.

The US is known for being a leader in agriculture and agriculture is the leading economic business sector in the economy of several states. However, over the past several decades, the number of farms has continued to shrink as individual farms become larger and larger and animal productivity continues to increase. For example, New York had 5,620 dairy farms in 2008, compared to 8,700 dairy farms in 2001, and 13,000 dairy farms in 1988, while the total number of cows being milked only decreased about 22% (from ~ 800,000 to 626,000 cows; NY has 6.7 of the total US cows). Aquaculture is still seen as a possible alternative agricultural enterprise in the USA, but remains as being a significant challenge to do so in a profitable manner. We believe that indoor aquaculture in particular offers an opportunity for food production in the USA, but must be approached with caution and by the prospective aquaculturalist doing their "homework" first. This book can be an excellent starting point for those considering commercial aquaculture or any form of aquaculture, even at the hobby level. Chapter 17 is an in depth discussion on the economics of aquaculture and some personal experiences from the authors. We hope you enjoy reading the rest of the book.

In this chapter, we'll review some basic background in aquaculture, some market realities, and future market opportunities. We'll conclude with some standard definitions and websites for further information. Finally, this book is an updated version of the original text we wrote several years ago (Timmons, et al., 2002). The "Yellow Book" has been extensively rewritten to reflect the latest information available from

research universities and commercial equipment suppliers. In addition, two new chapters on biofiltration and denitrification have been added and a consistent design example for an Omega Fish Commercial Production System. Some chapters remain largely the same, for example Chapter 3 Mass Balances, Loading Rates, and Fish Growth. Some things just never change!

1.1 THE OPTIMISTIC VIEW

Peter F. Drucker, a world recognized business leader and economic forecaster, predicts that aquaculture –the farming of aquatic organisms– will be one of three major economic opportunities in the new millennium. Everyone everywhere is either eating more fish or thinking they should! Changes in dietary patterns and the fact that the US Surgeon General now recommends eating fish as a significant protein source for the diet are strong indicators that opportunities in aquaculture will continue to expand. The US catfish industry is a ready example of how fast aquaculture fish markets can grow; i.e., this market grew by 100,000 tons (220 million lbs) in the mid 90's. The Chilean salmon industry has grown from $159 million industry in 1991 to exporting over $1.7 billion in 2005, and now employs 53,000 people. The production of tilapia has been exponential in the last several years to the point that the US market demand for tilapia has gone from essentially nothing to importing the equivalent of 270,000 tons (600 million lbs) in year 2005.

We believe that aquaculture is the most probable and feasible solution to providing the seafood products for this ever increasing market demand and shrinking supply of product from the oceans. Aquaculture is an environmentally responsible alternative to fishing. It provides a consistent and reliable source of high quality, fresh seafood that is nutritious, safe to eat, and can be reasonably priced.

1.2 RECIRCULATING AQUACULTURE SYSTEMS (RAS)

Fisheries products are the last mass marketed food being supplied to consumers by "hunter-gatherers". This method of bringing product to market is rapidly becoming obsolete, and is no longer able to meet current market needs. As a result, aquaculture is the fastest growing segment of agriculture, and is now supplying over half of all seafood consumed (see Table 1.1). Note that when the portion of wild catch used for animal feeds is removed (33% of total), aquaculture supplied seafood accounts for 45% of the total supply.

Table 1.1 Contributions from Wild Catch and Aquaculture (fisheries data from the FAO Fisheries Global Information System site, May 2010. www.fao.org/figis)

Production (Million Ton)	1950	1960	1970	1980	1990	2000	2007	2020 estimated
Wild Catch	19.2	34.7	63.7	68.2	85.9	96.8	99.3	129.8[A]
Aquaculture	0.6	2.0	3.5	7.3	16.8	45.7	55.4	103.2[B]
Total	19.8	36.7	67.2	75.5	102.7	142.5	154.7	233.0
% from Aquaculture	3%	5%	5%	10%	16%	32%	36%	44%
World Population (billions)	2.556	3.040	3.709	4.453	5.283	6.082	6.670	7.202
Per Capita Food Fish Supply, kg	5.2	8.0	12.1	11.3	12.9	15.6	17.0	17.1[C]

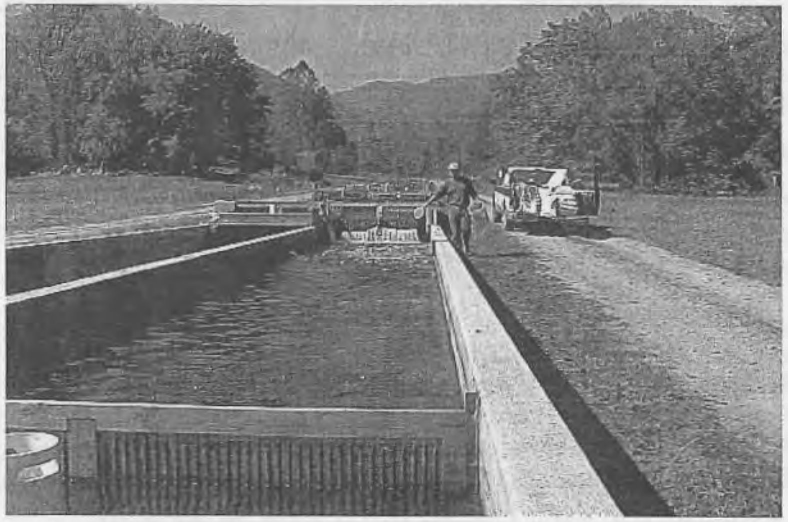
NB: Approximately 33% of the Capture Fish are converted to fish meal/oil
[A] Assumes 1.5% increase in capture fisheries production per year (Delgado et al. 2002)
[B] Assumes 2.8% increase in aquaculture production per year (Delgado et al. 2002)
[C] As projected by Delgado et al. (2002)

AQUACULTURE SYSTEMS

Seafood is only as good as the water in which it lives. Aquaculturists control the quality of the water, so the seafood they produce can be free of environmental contaminants. Consumers have demonstrated a marked preference for cultured/farmed seafood because it is more consistent in quality and presentation and it tends to have a milder taste than wild seafood.

Aquaculture systems can be extensive, semi-intensive, or intensive, depending upon the number of organisms grown per volume of water and the water source and supply. Pond culture is extensive, cage culture is semi-intensive but intensive within the cage, and RAS are intensive systems. Pond and cage systems are open-air, and therefore there is always a risk of air or water-borne contaminants. Because water quality control is more difficult in pond and cage systems, the number of

organisms that can be grown effectively is limited. In this book as noted by the title, we are concentrating on recirculating aquaculture systems (RAS). The principles of RAS for the water environment can be employed in the open air, but you lose total control of the environment.

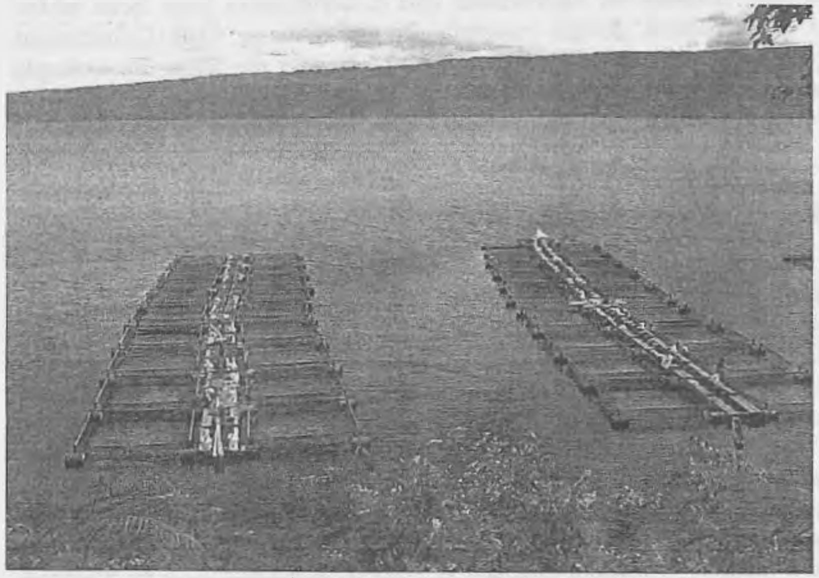


Typical Raceway type system

Conventional aquaculture methods, such as outdoor pond systems and net pen systems, are not sustainable in the long term, due to significant environmental issues and their inability to guarantee the safety of their products to the consumer. Conversely, indoor fish production using RAS is sustainable, infinitely expandable, environmentally compatible, and has the ability to guarantee both the safety and the quality of the fish produced throughout the year.

Outdoor pond (warm water systems, e.g., catfish) and net pen aquaculture systems (cool water, e.g., salmon) are disadvantaged by their:

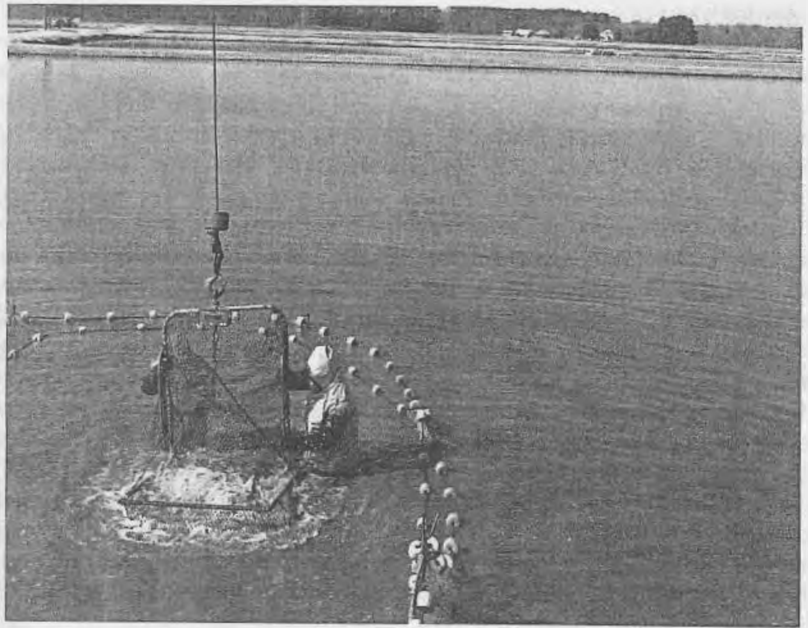
- Large footprint requirements
- Limited appropriate natural sites
- Environmental issues with respect to the management of the fish excrement
- Geographical limitations due to the need for a perfect growing climate
- Vulnerability to disease, predators, and natural disasters via their outdoor uncontrollable environment



Typical net-pen aquaculture

Outdoor pond and net pen-based systems are significantly disadvantaged with respect to the potential for disease, which could result in culture loss. Diseases in fish systems are transferred by direct water contact with diseased organisms. Indoor systems start with potable water and unless diseased fish or fish carrying diseases are introduced into the culture system, there is minimal potential for disease introduction. And if there is a disease event, effective treatment is much more manageable than what a fish culturalist faces with traditional outdoor systems.

The outdoor pond and net pen-based systems are also disadvantaged by their inability to supply a consistent product due to difficulties controlling the growing cycles, which then creates peaks and valleys of supply available to the market. Finally, issues related to escapement are of major concern: particularly with biotechnology modified species. In such cases, RAS becomes the only acceptable culture technique because the animals cannot escape an indoor RAS and will, therefore, not have any impact on the natural populations.



Typical pond aquaculture operation
(note levees in background to separate ponds)

1.3 RAS Advantages

Indoor RAS offers the advantage of raising fish in a controlled environment, permitting controlled product growth rates and predictable harvesting schedules. RAS conserve heat and water through water reuse after reconditioning by biological filtration using biofilters. RAS allow effective economies of scale, which results in the highest production per unit area and per unit worker of any aquaculture system. RAS are environmentally sustainable; they use 90-99% less water than conventional aquaculture systems; less than 1% of the land area; and provide for environmentally safe waste management treatment. Table 1.2 provides a comparison of water used per kg of fish produced. The RAS assumes a tilapia culture system with a density of 100 kg/m^3, a 1% feeding rate, and a feed conversion of 1 to 1 and a system volume discharge rate of 5% per day. Some current commercial RAS are using less water (2 or 3% system discharge per day and of course some use much more), higher densities and similar feed conversions. RAS allow year-round production of consistent volumes of product, and complete climate control of the environment. Because RAS can be set up to

produce the same volume of fish every week, week in and week out, they have a competitive advantage over outdoor tank and pond systems, which are seasonal and sporadic in harvest.

Table 1.2 Water and Land Use Per kg of Production of Aquaculture Products and a Relative Comparison to an Intensive RAS Tilapia Farm (RAS assumed to discharge 5% of system volume per day)

Species and System	Production Intensity (kg/ha/y)	Water required (Liter/kg)	Ratio of System's Land or Water Use to RAS Use: Land	Water
O. niloticus (Nile tilapia) **RAS produced**	1,340,000[a]	50	1	1
O. niloticus (Nile tilapia) **ponds**	17,400	21,000	77	420
I. punctatus (Channel catfish) **ponds**	3,000	3,000–5,000	448	80
S. gairdneri (Rainbow trout) **raceways**	150,000	210,000	9	4,200
Panaeid shrimp **pond** (Taiwan)	4,200–11,000	11,000–21,340	177	320

[a]does not account for land used external to building space

RAS designed aquaculture systems are infinitely scalable. There are no environmental limitations to the size of the intended fish farm to be built because waste streams are controllable in environmentally sustainable ways.

RAS offer a high degree of environmental control. This not only mitigates the risks of outdoor aquaculture (natural disaster, pollution, and disease) but also allows for optimized species growth on a year-round basis. A similar optimization can be observed in the domestic poultry industry, where chickens were brought indoors and the cost of environmental control was more than recovered by higher growth rates, improved feed conversion, and more efficient use of labor. This is demonstrated by the fact that broiler growers produce 1,000,000 kg of chicken per man-year of effort. In addition to the growth advantages afforded by RAS technology, the low environmental impact of these

systems means that they can be built closer to the consumer and replicated rapidly.

Indoor aquaculture is probably the only potential method that could be used to ensure a 100% safe source of seafood, free from all chemicals and heavy metals. With increasing consumer concerns about food safety, aquaculture producers using RAS have an unprecedented opportunity to meet the demands for safe seafood. Attributes of fresher, safer, and locally raised product are clear advantages for RAS produced seafood. Because RAS can be set up to produce the same volume of fish every week, week in and week out, these systems have a competitive marketing advantage over outdoor tank and pond systems, which are seasonal and subject to environmental disaster beyond the control of the operator.

WATER REQUIREMENTS, USE, AND CONSERVATION

Traditional intensive fish farming systems use flowing water resources for two purposes:

- to transport oxygen to the fish, and
- to carry the waste produced in the system (metabolic by-products and other materials) away so that they do not accumulate in/around the fish farm to undesirable levels.

More recently, the carrying capacity of a flowing water fish farm has also become limited by imposition of state or federal discharge regulations. For example, traditional trout culture requires relatively large volume water resources to produce fish in a single-pass tank and serial-reuse raceway system.

> **"RULE OF THUMB"**
>
> 50 lb production per gal/min of flow per annum
>
> (6 kg/yr per Lpm)

Clear Springs Trout Company (Buhl, Idaho), established in 1966, produced 10 million kilograms of trout in 2004, and is the world's single largest producer of rainbow trout for human consumption (personal communication, Randy MacMillan, Clear Springs Food Company). According to MacMillan (2006), Clear Springs non-consumptively uses 22.6 m^3/s > (360,000 gal/min) of water flow for up to five serial-reuses through concrete raceways. The only effluent treatment is either quiescent zone settling within the raceways or sometimes full-flow settling following the raceways. Low phosphorous feeds are used to help minimize effluent phosphorus and comply with NPDES permit requirements. MacMillan also said that depending on facility design, they produce 37 to 71 lb (17 to 32 kg) per gpm of flow per annum (4.5 to 8.5 kg per Lpm).

Fortunately, serial-reuse systems may not have to actually capture large percentages of waste solids for their average discharge to meet concentration-based effluent limits on TSS (total suspended solids), even in Idaho where permits issued by NPDES (National Pollution Discharge Elimination System-EPA promulgated) limit fish farm effluents to monthly average suspended solids concentration of 5 mg/L (net). Similarly, serial-reuse systems may not have to capture large percentages of waste solids under typical fish production levels, i.e., 50 lb production per year per 1 gal/min or 6 kg/yr per Lpm, because a simple mass balance shows that only about 5 mg/L of total suspended solids concentration would be added to the flow if averaged over the entire day. Also, nutrient limits (P, K, etc.) are becoming a discharge issue. However, because of the large water volumes used in single-pass and serial-reuse production systems, it is not realistic for farms such as Clear Springs Trout Company to remove nutrients from their effluents (IDEQ, 1998). Therefore, some treatment (often using settling basins) is used to ensure that spikes in TSS are not discharged, although overall waste capture efficiencies may only be 25–50% within serial-reuse systems (Mudrak, 1981); this topic is discussed in detail in Chapter 5 Solids Capture.

To abate the environmental impact of aquaculture, production practices and technologies are being adopted to minimize waste production, conserve water, and concentrate wastes into smaller flows during fish culture, thus the heightened interest in RAS. As mentioned above, traditional flowing water systems can produce approximately 6 kg of fish annually for every 1 Lpm of water flow. By reusing or recycling 80 to 90% of the water prior to discharge, partial-reuse systems can produce as much as 48 kg of fish annually for every 1 Lpm of make-up water flow, i.e., 400 lb of fish annually for every gallon per minute of water flow. And of course in the extreme case where a 100% RAS is employed, the production is based upon rates of evaporation which means that several 100 fold increases in production per unit of water can be achieved compared to flow through production.

Fully recirculating systems, because of their extremely low makeup water requirements, can readily capture from 96% (Heinen et al. 1996) to 100% of the waste produced, depending upon the percentage of make-up water passed through the system. In comparison, a well operated serial-reuse raceway system can typically achieve overall waste capture efficiencies of only 25–50% (Mudrak, 1981). Additionally, by using "Cornell-type" dual-drain circular culture tanks and either bead filters or microscreen filters, recirculating systems can produce a much smaller and more concentrated waste stream, which can be treated more

economically and efficiently (Timmons et al. 1998; Summerfelt et al. 2000A; 2000B Summerfelt, 1996; 1999). Thus, partial-reuse and fully-recirculating systems offer key advantages over traditional fish culture in serial-reuse raceway systems, including 80–100% reduced water resource requirements (respectively) and an overall waste capture efficiency of 80–100% (respectively). Also, solids removal from dual-drain circular tanks is so rapid and effective that partial-reuse and fully-recirculating systems can treat and return water to the culture tanks with <2.0 mg/L TSS (Summerfelt et al. 2000A; 2000B). Coldwater recirculating technology has now advanced to the point that these systems can provide a controlled environment with optimized water temperature, quality, and culture tank velocities (Summerfelt, 1996; Summerfelt et al. 2000A; Summerfelt et al. 2001).

1.4 World Market Needs

Aquaculture must continue to increase its capacity as the wild catch is predicted to only marginally increase by 1.5% yearly over the next 15 years (Delgado et al. 2002), which will not supply the needed demand. In addition, the public is increasingly demanding production of seafood to be from sustainable methods that are eco-friendly. Our basic thesis is that RAS are the key technology that will allow the world aquaculture community to supply the world per capita needs in seafood over the next decade and to do so in an environmentally friendly manner (see earlier Table 1.2 for RAS use of water and land compared to other competing forms of aquaculture).

RAS produced supply should not be thought of as competing with the wild catch, but of complimenting the ability of the oceans to supply some sustainable level of production that must be supplemented by aquaculture produced product. This is not a contest between traditional fishing methods and new technology. An additional 77 billion kg of supply is needed by the year 2020 to maintain current world per capita consumption levels. We predict that 44 billion kg of this demand will be met through aquaculture.

The recognized deficit in fishery supplied (wild catch) product has led to concerted efforts to better manage natural fisheries. For the case of United States and likely appropriate to other countries, Macinko and Bromley (2002) argue that policy makers must recognize that the American (or country of interest) public owns the nation's fisheries. Poor management of fishery resources is at the heart of the fishery crisis. The

US (or country of interest) should manage its fisheries as it manages its other natural resources

Macinko and Bromley acknowledge that managing through an individual fishing quota approach, IFQ; will only ameliorate the race for the 'last' fish. Even with improved management of the ocean resources, to maintain similar world per capita consumption levels as in 2005 will require an increase in production from aquaculture of 44 million ton[1] by the year 2020 (assumes an increase per year of 1.5% in the capture fisheries and a 2.8% increase in aquaculture, Delgado et al., 2002).

1.5 Market Dynamics

With the fishing industry suffering from dramatic reductions in the supply of wild caught traditional species, alternatives to wild catches now have a competitive advantage in the marketplace. Where aquaculture was once viewed as the wave of the future, it is now generally accepted as a significant source of product. In 2009, more than half of all the seafood consumed around the world was a product of aquaculture.

The leading forms of dietary protein consumed in the US in 2007 ranked as follows (all expressed on a per capita basis; USDA, 2009; see http://www.ers.usda.gov/Data/FoodConsumption/FoodAvailSpreadsheets.htm#mtpcc):

- Beef 65.1 lb (29.6 kg)
- Chicken 85.4 lb (38.8 kg)
- Pork 50.3 lb (22.9 kg)
- Turkey 17.5 (8.0 kg)
- Seafood 16.3 lb (7.4 kg)

Inconsistent supply and subsequent higher retail costs has kept US consumption of seafood steady during the 1990's at around 14 to 15 pounds (7 kg) per capita. However, since 2000, there has been a slow but steady increase from 15.2 lb (6.9 kg) in 2000 to 16.53 lb (7.5 kg) in 2006, which represents over 400 million lb (180,000 ton) increase in product supply. The wild catch is simply not keeping up with the demand by the consumers, and as a result, the supply of aquacultured seafood products has risen to meet the demand. Due to the highly favorable opinion of seafood (most Americans view seafood as being healthy); it is

[1] This book will use ton as 1,000 kg

thought that the increased supply of cost-effective aquacultured products may increase overall seafood consumption rates. For example, the tilapia market is growing because consumers find its flavor to be delicate and its texture flaky compared to other species at a time when reduction in the supply of wild caught traditional species such as cod, haddock, halibut, and pollock, is forcing utilization of other species. Tilapia are of particular interest because they adapt readily to RAS and almost all of the US production of tilapia is from RAS, the subject of this book.

United States per capita consumption rates of individual seafood species is presented in Table 1.3. As indicated in bold typeface, ***aquaculture*** seafood species (shrimp, salmon, catfish and shellfish) are becoming a significant portion of the $8.8 billion United States seafood market and have increased market share in contrast to ocean caught species such as cod.

Table 1.3 Current U.S. Seafood Consumption Rates by Species in Pounds (Bold signifies partial contribution from aquaculture products, source National Marine Fisheries Institute, 2009)

Species	2008 Rank	2008 lbs	2005 lbs	2000 lbs	1995 lbs
Shrimp	**1**	4.10	4.10	3.20	2.50
Canned Tuna	2	2.80	3.10	3.50	3.40
Salmon	3	1.84	2.43	1.58	1.19
Pollock	4	1.34	1.47	1.59	1.52
Tilapia*	**5**	1.19	0.85	NR	NR
Catfish	6	0.92	1.03	1.00	0.86
Crabs	7	0.61	0.64	0.38	0.32
Cod	8	0.44	0.57	0.75	0.98
Clams	9	0.43	0.44	0.47	0.57
Flatfish	10	0.43	0.37	0.42	0.30

* Fillet basis (yield is~ 30% on tilapia)

The authors predict that the US tilapia industry will soon surpass the catfish industry and that eventually, RAS produced tilapia will be comparable to the US broiler industry that produces 15 billion lbs (6.8 million ton) per year of chicken. This is because RAS will continue to become more cost efficient, and RAS technology allows production to be infinitely expanded. Generally, the primary constraint to building a fish farm or other animal production unit is finding a site that can handle the waste disposal needs. Natural environments that will support traditional aquaculture such as raceways, net-pens, and outdoor ponds are extremely limited and subject to a political process. For example, in Canada recently, a net pen operation was denied their site renewal permit without explanation after 10 years of trouble free operation. RAS technology has a primary and exclusive advantage by minimizing the effluent waste stream by a factor of 500 to 1,000 or simply no discharge! Even so, RAS sites still need to address the issue of waste disposal, but at the reduced effluent load and ability to concentrate the waste stream, there are many cost effective waste treatment options, Chapter 6.

Another key advantage of using RAS is that these production sites can be located near the markets for the products being produced. Fresh product becomes the immediate focus, since *fresh seafood often commands twice the product value* of frozen seafood. There is a strong consumer preference for fresh and RAS are ideally suited to provide this product. Locating near the market also minimizes product-hauling costs and maximizes available shelf life. For reference only, Table 1.4 provides a breakdown of current US consumer preference and wholesale pricing on fresh seafood:

Table 1.4 US Market for Fresh Seafood (Urner-Barry and New England Fish Exchange Auction, August 2006)

Per Capita Consumption by Species	lb/yr	(kg/yr)	Wholesale Price, $/lb
Tuna-Yellow Tale	3.30	1.50	$6.95
Shrimp	4.20	1.91	
Alaska Pollock	1.28	0.58	$0.52
Salmon, Atlantic 4-5 lb	2.15	0.98	$4.40
Cod, head off	0.63	0.29	$3.37
Cod, boneless			$5.50
Catfish (price is for fillets)	1.09	0.50	
Clams	0.47	0.21	
Crabs	0.63	0.29	
Flounder/Sole (Flatfish)	0.33	0.15	$2.12
Scallops	0.28	0.13	$6.75

1.6 Overview of Recirculating Aquaculture Systems (RAS)

Fish can be stocked most intensively in RAS. In RAS, the total environment is controlled. Fish are raised in tanks and in the most secure environment are placed within an enclosed building to control the aerial

environment as well. Water circulates throughout the system, and only a small percentage of the water is discharged daily, e.g., 10% per day or less of system volume. Temperature, salinity, pH, alkalinity, chemical composition, and oxygen are all monitored and continuously controlled. Solid wastes are filtered and removed, oxygen is added to maintain sufficient dissolved oxygen levels for the stocking density, and effluent water is passed through a biofilter for a biological conversion of ammonia-nitrogen to nitrate-nitrogen. Designing and operating RAS requires a solid understanding of the many unit processes or operations involved:

- Mass balances (see Chapter 3)
- Culture Units (see Chapter 4)
- Solids Capture (see Chapter 5)
- Nitrification (see Chapter 7)
- Gas Transfer (see Chapter 10)
- Fluid mechanics (see Chapter 12)
- Waste management (see Chapter 6)
- Feeds and nutrition (see Chapter 18)
- Biosecurity (see Chapter 16)
- Systems monitoring (see Chapter 13)
- Building heat and moisture control (see Chapter 14)

The failure of any one of these operations can cause the whole system to fail, usually killing the fish in the process. These operations will be covered in detail in later chapters of this text.

RAS are more capital-intensive than most other types of traditional aquaculture systems, and must rely on economic productivity per unit volume of rearing space for profitability. An RAS farm will have support features such as backup generators whose cost is also part of the overall production costs for the farm. Sufficient production quantity must be achieved in order that these support component costs are reduced per unit of production (e.g. per kg produced) to a level that the overall production costs are competitive. Large production levels are achieved by replicating a successful modular unit that insulates itself from failure of other production modules. This feature permits controlled, incremental increases of seafood production in response to rising market demand. It also facilitates raising capital and spreading investment risk through business arrangements such as franchising, contract growing, and production/marketing cooperatives. Finally, RAS technology is species-adaptable, allowing operators to follow market trends for seafood preference.

1.7 CAN RAS COMPETE

RAS are now being successfully used to raise tilapia in northern US climates at costs of less than $2.20 per kg (whole fish basis) and outdoor intensive production in Central America has reduced costs to the $1.00-$1.30/kg range (see Table 1.5).

Table 1.5 Costs of Tilapia Production in Selected Countries (2002 personal communication, Dr. Ragnar Johannsen, Rannsóknastofnun Fiskiðnaðarins, Reykjavík, Iceland)

COUNTRY	Cost of Production	
	$/lb	$/kg
Brasil, Ecuador, Cuba	$0.50	$1.10/kg
Costa Rica, Jamaica	$0.55	$1.20/kg
Colombia, México	$0.68	$1.50/kg
USA	$0.91	$2.00/kg

The proportional costs that make up production are summarized in Table 1.6 for a current commercial tilapia operation in the Northeast US producing 450,000 kg per year (feed at $0.55/kg, electric rates are $0.03/kwh, gas heating at $0.0085/MJ, and oxygen at $0.09/kg delivered). The perception that heating and pumping costs make RAS-produced fish that are excessively high in cost is false as shown in Table 1.6 where these costs are only 15% (9% heating and 6% electrical/pumping) of the total direct costs of production. Note though that local utility costs or price escalations in fuels and energy can affect the competitiveness of a specific location.

Table 1.6 Representative Cost of Goods Sold (COGS) Expressed as Component Costs to Produce Tilapia (water & sewer supplied by a public utility)

Cost of Goods Sold	% of Total
Purchases - Fish Stock	7%
Purchases - Feed	28%
Purchases - Oxygen	13%
Direct Labor	29%
Supplies	3%
Utilities	
natural gas	9%
electricity	6%
water & sewer	5%
Product Delivery Costs	1%
Total COGS	100%

Currently there are no commercial recirculating aquaculture operations in the US that have sufficient scales of production or processing to compete with large scale aquaculture or off-shore commercial fishing operations in the food service markets or wholesale [2]. To be competitive with the major non-US tilapia producers, a farm would need to produce a minimum of 3 to 4 million kg/yr (3,000 to 4,000 ton) of annual production to justify an automated processing plant and have direct production costs of \$1.10/kg or less preferably. This is a challenge and realistically only those extremely well-managed farms that have energy site advantages (free heat and low cost electricity < \$0.04/kWh) could expect to do this.

[2] At the time of writing, there were several tilapia facilities planned for the US locations that were in excess of 5 thousand ton capacity.

COMPETING MEAT PRODUCTS

Aquaculture products however they are produced and at whatever scale must ultimately compete against other choices of meat proteins. Table 1.7 shows the quantities of meat consumed since 1960 in the US (USDA Economic and Research Service, 2009).

Table 1.7 Per Capita US Consumption (kg) of Various Meat Products* 1960 to 2007 (Delmarva Poultry Industry, 2009 & Economic Research Service/USDA, 2009)

Year	Beef	Pork	Total Red Meat	Broilers	Turkey	Total Poultry	Total Red Meat & Poultry	Commercial Fish & Shellfish
1960	28.7	26.8	59.7	10.7	2.9	15.6	75.3	4.7
1970	38.4	25.3	66.2	16.6	3.7	22.0	88.2	5.4
1980	34.8	26.0	62.1	20.8	4.7	26.5	88.6	5.7
1990	30.8	22.6	54.5	27.0	8.0	35.9	90.4	6.8
2000	28.5	23.2	54.8	34.9	7.9	43.3	98.1	6.9
2007	30.9	23.2	54.9	38.5	7.9	46.4	101.3	7.4

* All products on a retail weight basis, except "other chicken" and "turkey" which are reported by USDA on a carcass-weight basis.

The broiler industry has shown a steady increase in per capita consumption over the last 45 years. Overall meat consumption has risen from 75 to 101.3 kg per capita and at the same time the US population has increased from 181 to 305 million people. Growth in the early years of the industry exceeded 20% per year and has continued to the present day where an annual growth rate of approximately 5% is being sustained (see Figure 1.1). Conversely, beef consumption peaked at 42.9 kg per capita in 1976 and has dropped to the current level of 30.9 kg per capita in 2005 (an increase from 2001 from 30.1 kg). Based upon current population levels, this is a loss of 12 kg per capita or 3.6 billion kg of product for the entire US population. If seafood is to exert a similar influence in diverting meat consumption, then the only way to do this is via market price. The aging population and the accepted health benefits of eating seafood should drive the consumption curves, given price competitiveness. To make dramatic changes as was exerted by the poultry industry, the market price of seafood must be less than competing quality meats. The market share for expensive protein, the current

condition for seafood, is essentially maximized at around 7.4 kg per capita in the US.

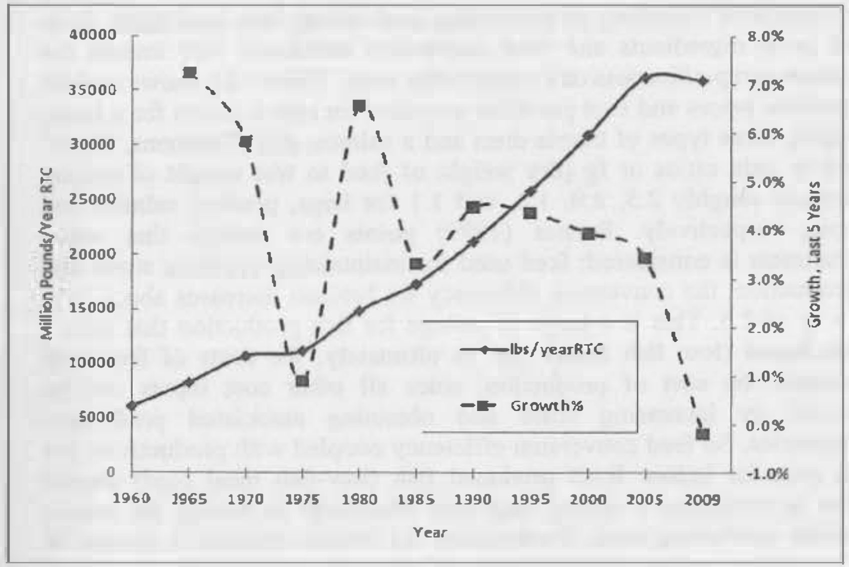


Figure 1.1 Growth of US broiler production from 1930 to 2009 in millions of pounds ready to cook (RTC) weight (left ordinate axis) and compounded growth rate for preceding years (right ordinate axis).

Hicks and Holder (2001) reviewed salmon production costs for net pen operations that produce on the average 1.36 million kg per cycle of 18 months. Salmon farming continues to increase in scale to further reduce costs of production. Productivity per person for a net pen operation is 136,000 to 204,000 kg per person per year. The present workload per net pen farm is about five individuals that care for a placement of 500,000 smolt that will be reared to a market size of 4 kg with a culling and mortality down to 400,000 animals. This translates into 213 ton per person per year. In addition, there are net changing crews and divers, so the support would be about 1-2 additional full time people. Thus, it requires a maximum of 7 people, which reduces the net productivity for a net pen operation to 152 ton per person. This is why the scale is being driven to even larger placements of smolts, so that the labor cost per unit of meat produced is further reduced. Some salmon operations are basically doubling the stocking numbers per site using

almost the same number of people to manage the site, making productivity 450,000 kg/year per full-time equivalent (FTE) worker.

Table 1.8 Comparison Of Production Costs ($/Kg) for Net Pen Salmon (Current and Most Efficient Operations), Large Scale RAS Produced Tilapia, and Commercial Broiler Production (Tilapia And Broiler Figures are from Timmons et al., 2002)

	Cost/kg			
	Tilapia	Salmon	Efficient Salmon	Broilers
Costs of Goods Sold (COGS)				
Direct Labor & Benefits	$0.17	$0.20		
Feed	$0.46	$1.26		
Oxygen	$0.11	$0.00		
Other Operating Costs	$0.04	$0.31		
Utilities - Heat	$0.22	$0.00		
Utilities - Electric	$0.09	$0.00		
Fingerlings	$0.18	$0.35		
Insurance	$0.00	$0.11		
Health Treatments	$0.00	$0.02		
COGS ($/kg fish produced)	$1.27	$2.25	$1.76	$0.66

Using the Hicks and Holder numbers, a comparison is made in Table 1.8 among:

- Large-scale tilapia RAS production
- Current most efficient salmon producers
- Broiler production

Although broiler costs of production are relatively low compared to the other fish products, the percent breast meat from a whole chicken is only 15%. Using 15% breast meat yield and similar other prices for packaging etc, Table 1.9 summarizes the FOB costs for the premium flesh pieces of an animal carcass.

Table 1.9 Premium FOB Prices ($/Kg) For Fish Fillets and Broiler Breast Based Upon Costs of Goods Sold (COGS); Assumes Fillet Yield of 31% Tilapia, 50% Salmon and 15% Broiler Breast Meat

	Cost $/kg			
	Tilapia	Salmon	Efficient Salmon	Broilers
COGS ($/kg fish produced)	$1.27	$2.25	$1.76	$0.66
Finance Operations	$0.07	$0.07	$0.07	$0.07
Finance Capital	$0.04	$0.04	$0.04	$0.04
Depreciation	$0.09	$0.07	$0.07	$0.07
Harvesting/Processing/Packaging	$0.55	$0.55	$0.55	$0.55
Total Cost of Production ($/kg)	$2.02	$2.97	$2.49	$1.39
Cost per kg dressed weight (83% yield)	$2.43	$3.58	$3.00	$1.67
Fillet cost per kg (31% tilapia & 50% salmon fillet yields)	$6.52	$5.95	$4.98	$9.25

The relatively low percentage yield for broiler breast meat has been a distinct disadvantage for the broiler industry. This has been addressed by steadily progressing to a further processed product (see Table 1.10). It should be no surprise to the seafood industry that marketing whole fish products has no future if the broiler industry can be used as any guide to consumer preference and as a means to add value to carcass parts that are not readily marketed.

Table 1.10 Product Form (% basis) for Marketing Broiler Products (Source: www.dpichicken.org/faq_facts/)

Year	Whole Bird	Cut-up	Further Processed
1962	83	15	2
1965	78	19	3
1970	70	26	4
1975	61	32	7
1980	50	40	10
1985	29	53	17
1990	18	56	26
1995	11	53	36
2000	9	46	45
2003	8	42	50
2009	11	41	48

Feed Cost Advantage for Aquaculture

Being competitive long term in the commodity meat market will depend to a large degree on the cost of feed used to grow the animals and the associated efficiency of converting feed energy into meat flesh. Both feed price ingredients and feed conversion efficiency will impact the ultimate competitiveness of a commodity meat. Table 1.11 shows current ingredient prices and cost per Kcal provided for ration mixes for a hogs, broilers, three types of tilapia diets and a salmon diet (Timmons, 2005). Feed to gain ratios or fg (dry weight of feed to wet weight of animal gain) are roughly 2.5, 2.0, 1.2, and 1.1 for hogs, poultry, salmon and tilapia, respectively. Forster (1999) points out though that when recruitment is considered; feed used for maintaining breeding stock and reproduction, the conversion efficiency for broilers increases about 25% or a fg of 2.5. This is a large advantage for fish production that uses a plant based (low fish meal) diet as ultimately, the costs of feed will dominate the cost of production, since all other cost inputs can be reduced by increasing scale and obtaining associated production efficiencies. So feed conversion efficiency coupled with productivity per unit area for indoor RAS produced fish (low-fish meal feed) should allow to command a strong long-term advantage in having the lowest possible production costs. Productivity for broiler chickens is around 76 kg per square meter of building space per year and indoor warm-water fish systems using 2.4 m deep tanks will produce around 290 kg per square meter of building floor area (assuming tank space is 40% of floor coverage).

Table 1.11 Relative Cost of Feed for Various Commodity Animals

Component	Cost $/ton	Hog	Broiler	Tilapia	Salmon
Protein		16%	24%	36%	55%
ME of diet, Kcal/kg		3,465	3,300	2,800	4,400
Fat (bulk)	$570	6%	6%		
Corn	$148	70%	59%	15%	
Soy (48%)	$335	23%	30%	52%	20%
Wheat	$177			20%	
Fish Meal (62% Protein)	$1537		2.5%	10%	50%
Fish Oil	$981			2%	28%
Blended Ingredient Cost $/1000 Kcal		.034	.044	.080	.139
Blended Ingredient Cost $/ton		$215	$264	$405	$1110

GENETIC IMPROVEMENTS IN AQUACULTURE

If we can be an optimistic projectionist for discussion sake, the information in Table 1.11 is very exciting for the potential of tilapia production in the US. Current US tilapia grower diets at 36% protein would be easily ~ $560/ton or 3 times the blended ingredient cost. Broiler diets will run $180 per ton or only about 1.3 times the blended ingredient cost. Now, if the US tilapia industry achieved a large scale industry, then one could expect similar economies of scale on the feed costs supplied to the farm, or something more like $240/ton. This would cut the cost of the feed component by over half compared to current costs in this undeveloped US industry. At some point, the US might actually start trying to capture some of the 500,000 ton (whole fish equivalent) US tilapia market that is being supplied by non-US producers. Even more optimism can be generated if one could anticipate improvements in the genetic performance of the current tilapia strains. As an example of what can happen when universities and industry cooperate is the steady improvements in broiler performance over the years (see Table 1.12). Growth rates, feed conversion efficiency, and mortality parameters have all improved by at least factors of 2. Imagine if such improvements could be realized in the tilapia industry. An industry is waiting to be born.

Table 1.12 US Broiler Performance from 1925 to 2009 (National Chicken Council)

Year	Market Age Average days	Market Weight kg, live weight	Feed to Meat Gain kg of feed to kg of broiler, live weight	Mortality Percent
1925	112	1.14	4.70	18
1940	85	1.31	4.00	12
1950	70	1.40	3.00	10
1960	63	1.52	2.50	6
1970	56	1.64	2.25	5
1980	53	1.78	2.05	5
1990	48	1.98	2.00	5
2000	46	2.28	1.95	5
2009	47	2.56	1.92	4

1.8 IS AQUACULTURE FOR YOU

There seems to be an assumption that given alternate forms of animal husbandry, a currently unsuccessful dairy or hog farmer could become a

successful fish farmer. Let us call that General Falsehood #1. Fish farming and RAS aquaculture in particular are generally more critically dependent upon expert management and precise activity than are other forms of farming. Thus, it is unlikely that a farmer with "average" management skills would be successful in fish farming. On the other hand, a successful dairy farmer could probably be a successful user of RAS technology. The importance of good management to a successful RAS farm cannot be overemphasized. Poor management is almost always the primary reason for failure in aquaculture ventures. Experience with flow-through raceways or outdoor ponds has almost nothing to do with understanding how to effectively and efficiently manage an RAS farm.

"General Falsehood #1"
A currently unsuccessful dairy or hog farmer could become a successful fish farmer

1.9 SOME QUICK CASE HISTORIES

The authors have been involved in several RAS startup ventures in their public institution and private roles. Some have resulted in failure. Failure is valuable in that it accentuates problems that must be recognized and addressed. The senior author led two Cornell University technology transfer efforts that were unsuccessful. Later in 1997, he started a private large tilapia farm that was still operating in 2006 and was producing over 500 tons per year of tilapia. (Chapter 17 provides details on this operation). The first technology transfer failure (mid 1980's) involved a very successful integrated turkey farm. This business had 130 employees, a processing plant, an extensive distribution network for fresh and frozen turkey products, and a restaurant. The second failure in the early 1990's was a cooperative trout farm that consisted of seven members named the Northern Fresh Fish Cooperative (NFFC). The cooperative members were smart, talented, experienced, and successful people – all with college degrees and some with advanced degrees. Failure of the first two efforts had the following common elements:

- Individuals had no previous experience with indoor fish culture
- Technology that was labor intensive
- Engineering Technology was poor
- Sensitive species (brook and rainbow trout)
- Enterprise was essentially an expensive hobby venture

Cornell designed the aquaculture system that was implemented at the turkey farm and assisted them with their implementation: four 10,000 gallon (38 m^3) culture tanks designed for 0.7 lbs per gallon (84 kg/m^3) at harvest density. The turkey farm ran their trout operation for 2 years before abandoning the effort. Solids accumulation was probably the primary reason for failure, even though there was a reasonably adequate solids settling chamber in the system. The turkey farm trout failure and subsequent analysis led the Cornell efforts to focus on the development of new biofilter designs and more energy efficient methods to move water between system components. Several years were spent refining the application of rotating biological contactors (RBC's), which replaced the submerged rock filter designs, and the use of airlift pumps to move the water between system components.

The efforts with the Northern Fresh Fish Cooperative including layouts of their system components are described by Timmons et al. (1993). Again, this particular system design had been successfully operated by Cornell for three years before the technology transfer effort and had achieved high carrying capacity (0.7 lb/gal, 84 kg/m^3), excellent feed conversions (1 to 1) and high fish growth rate (an inch per month, 25 mm/month). This system was quite amazing and efficient in that the airlift pumps moved water from the culture tank to the solids settling chamber to the biofiltration chamber and back to the culture tank on a total water level differential of 1.5 cm. The system also employed foam fractionators (Weeks et al. 1992; Chen et al. 1992; Chen et al. 1993, Chen et al. 1994 a, b; c; Timmons et al. 1995) in the culture tank to remove fine solids, provide oxygen and CO_2 removal, and increase the circular motion in the culture tank to assist in solids removal.

1.10 HISTORY LESSON ON FAILURES

In 1992, Peter Redmayne (Editor of Seafood Leader, January/February issue 1992) did a story on the financial failures in the aquaculture industry. He mentioned several notable failures and their reason for demise:

- Idaho-based J.R. Simplot Co. closed the doors on its two-year old, intensive tilapia operation, losing more than $20 million in the process. Reason: inadequate biofilter.

- Bodega Farms shut down its $9.5 million steelhead, coho salmon and abalone farm near Bodega Bay, CA. Reason: State of CA

would not allow two million fingerlings across the border, and they had no place else to go (no fish, no cash).

- Aquaculture Technologies of Louisiana (ATL) went bankrupt leaving 2,000 acres of catfish ponds in St. Landry Parish and $9 million in debts to some 300 creditors. Reason: bad management.

- NAIAD Corp, largest catfish farming and processing venture in Texas filed Chapter 11 (August of 1991) after starting processing its own catfish. Reason: lack of operating cash related to poor cash flow management.

- Blue Ridge Fisheries (Martinsville, VA) the largest indoor catfish operation in the world (at that time, 1991) lost its assets to bank foreclosure. Reason: the RAS was not cost effective (this facility was resurrected as a tilapia facility and is currently the largest producer of tilapia in the US (in excess of 2,000 ton), essentially under the same management structure).

In the 1990's, the following scenarios unfolded:

- Fish & Dakota lost several hundred tons of fish and did not reopen its doors. Reason: New management eliminated some of the 24 hour coverage and a power outage and failure of the "automatic" stand-by generator killed the fish.

- Sunflower Aquaculture (Kansas) lost their fish (not the enterprise) when their roof caved in; this was a military facility that had been unused for many years and had clear span timber trusses; when the trusses regained some moisture, they structurally failed.

- Northern Fresh Fish Cooperative (central NY), last member of the cooperative gave up when he lost all his trout because his dialer had not been hooked up to alert him of a lack of water (had left a drain open during a cleaning operation); the 2nd to last member went out of business when his well went dry.

- Perch operation in Western Pennsylvania closed their doors when their new system had finally reached design carrying capacity and then the liner in their culture vessel "broke".

- Southern Pennsylvania perch grower finally gave up after their initial stocking of perch showed growth rates a fraction of what was anticipated. They kept open their basil operation that was on the same property.

Be careful. Remember the following rule of thumb.

"Rule of Thumb"
Only invest what you can afford to LOSE!

The history lesson above should tell you something. The early aquaculture ventures (1980's) were large scale, since on paper there were large profits to be made and going large scale maximized profits. The moral of the story here is that the authors know of *no successful operation in excess of 200 ton/year that did not start out by building and operating a much smaller scale operation.* Mid 90's saw a steady influx of individuals trying to launch aquaculture operations. The theme during this period was "high value" species, as in, we know we can not compete using RAS in the major market fish species, but we can produce such-and-such and sell them for $40/kg. *This is a total myth.* You must be good and be extremely efficient at whatever species you are growing. If a particular species is retailing at high values, there is a reason.

In the 2000 era, we are seeing people being much more cautious about entering the aquaculture arena. Being in aquaculture does not mean you have to raise the fish you sell. Advice we often give is to buy fish from someone else and then develop a market for your product based upon service. If you can be successful here first, then you have a good chance of being even more successful if you raise your own product and control the supply chain over one additional step. This is further discussed in the next section. Read this next section carefully before deciding upon what role you want to play in the aquaculture industry, particularly for your first steps.

1.11 Interactions Between Objectives, Resources, Business Strategy, and Design

The authors frequently receive calls from individuals who desire to enter aquaculture with the callers having the inherent assumption that "aquaculture" means growing fish. And of course, everyone's objective is to have a profitable enterprise. The economics of RAS are thoroughly reviewed in Chapter 17 and the reader is encouraged to read this chapter

before starting an aquaculture venture. But, the first rule in RAS aquaculture is to never invest more than you can afford to lose. There are many hard-luck stories for every tale of success. Some families have lost everything in their pursuit of something they just wanted to do. One thing is certain: poor skills from one profession do not become great skills in aquaculture. You have to be good at it to make a profit and this generally entails several years of bands-on experience. This book gives you a head start, but is no substitute for real experience.

There are countless ways to become involved in aquaculture and perhaps your last choice should be to actually trying to raise fish (there are lots of people who have fish they want to sell and are not willing to do the marketing and support of a customer base). You should review your own short term and long term objectives, the natural resources available to the enterprise, the management and business skills available for this enterprise (referred to as business strategy), and finally the design of a system to achieve these objectives. If any one of these steps is ignored or components are missing or insufficient, then the success of the planned operation is unlikely. Design of a physical system should be the last step in the planning process.

Depending on the reader's own perspective, design is constrained by objectives, resources, and business strategy or the design expresses the object resulting from considering them. Although there is interaction between all components, the objectives must be determined first, and the resources available to pursue the objectives must be identified.

Table 1.13 lists some simple examples that will help to convey our thoughts behind the interactions among the resources, business strategy, and resulting design. The objectives for each case are similar and not specific enough to define a design other than stating a possible structure. Selecting different management strategies that result in alternate designs, as shown in the second and third case, may approach the identical objective. The same resources are given for each case and are again too broad to be of value in design but show dependency upon components. For example, a person need not possess water as a personal resource in order to work in the aquaculture industry. Why not consider buying fish from a grower who is not interested in marketing their product? These examples show that objectives, resources, and strategy must be specific to affect an appropriate design.

Table 1.13 Example Interactions of Objectives, Resources, Strategy and Design

Objectives	Resources	Business Strategy	Design
Have an annual income of $50,000	$500,000; financial institutions, water, market, knowledge	Invest at a net profit of 10%	Not necessary
Have an annual income of $50,000 from Aquaculture	Same	Buy and Sell	Not necessary
Same	Same	Buy, hold briefly & sell	Holding tanks, transport capability
Have an annual income of $50,000 from buying, processing tilapia fillets	Same	Buy, process, & sell	Processing facility
Have an annual income of $50,000 from selling tilapia fingerlings	Same	Maintain brood stock, incubate and hatch eggs, and maintain hatchery	Breeding tanks, egg incubator, fingerling production
Produce 114,000 kg per year of tilapia	Same	Maintain advanced growout system (needs more specific objectives, harvest size)	RAS or pond

1.12 TERMINOLOGY AND NOMENCLATURE

A primary requirement for discussing RAS technology is a common base of terms. There is a fairly well accepted terminology that is now in use among the aquaculture community, having been derived from a report filed the European Inland Fisheries Advisory Commission in 1987. The following are the definitions and terminology that will be used

throughout the chapters to come. Commonly used conversion factors are provided in the Appendix.

- **Carrying Capacity**: The maximum mass of aquacultured product that can be maintained within a culture system; usually expressed as mass per unit volume of the culture system
- **Flow Through Rate**: The volume of new water per unit time passing through a culture tank; refers to the make-up water specifically.
- **Mean Hydraulic Residence Time (HRT)**: Refers to the time required at a given rate of flow for a complete volume of water in a tank to be exchanged, V (volume of tank) / Q(flow rate)
- **Percentage Replacement**: The percentage of the total system volume replaced per day
- **Percentage Recycle**: The percentage of the total system volume that is retained, on a daily basis
- **Production to Capacity Ratio (P/C ratio)**: This is the ratio of system output per year to the maximum carrying capacity of the system. P/C ratios of 3 are typical of high density systems for fast growing fish.
- **Reuse (serial reuse)**: Water is reused in multiple tanks, moving in one direction **never used in the same tank twice (as NOT recycled)**; often referred to as serial reuse
- **Recycle (Recirculating Aquaculture Systems or RAS)**: Water flows from a tank (s) to a treatment process and then is returned to the tank, hence the term recirculated or recirculating aquaculture systems or RAS. RAS are generally regarded as systems that discharge less than 20-50% of the standing water in the system volume per day. Some RAS may discharge more water, but become increasingly more like a flow through system.
- **Specific Surface Area**: Surface area of the media per unit volume; usually referring to the surface area of a particular media used in filtration or settling components

- **Stocking Density**: Mass of cultured product per volume of tank (ignores the effects of fish displacing part of the water volume)

- **Total Biomass**: Mass of cultured product in the culture system

- **Total System Volume**: Volume of water in the culture tank, pipes, reservoirs, treatment tanks, and pumps

1.13 WEBSITES FOR REFERENCE

The reader is also referred to the following list of websites that contain vast amounts of information that can be useful for both the new and experienced aquaculturalist:

Alternative Farming Systems Information Center (AFSIC)
http://www.nal.usda.gov/afsic/
The AFSIC is one of 10 information centers at the National Agricultural Library. AFSIC serves as a national clearinghouse for aquaculture information and provides materials for aqua farmers, consumers, industry personnel, educators, government agencies, associations, libraries, the media, students, scientists, and prospective farmers.

Aquaculture without Frontiers
http://www.aquaculturewithoutfrontiers.org/
Aquaculture without Frontiers (AwF) is an independent non-profit organization that promotes and supports responsible and sustainable aquaculture and the alleviation of poverty by improving livelihoods in developing countries. Formed in 2004, AwF is registered as a charity in the UK and as a non-profit organization in the USA. AwF has been established for the specific purpose of promoting and supporting responsible and sustainable aquaculture to assist in poverty alleviation through improving rural livelihoods in developing and transition countries. In its work, AwF draws on the experience of respected professionals from every relevant discipline. AwF already has a database of more than 80 volunteers.

U.S. Trout Farmers Association
http://www.ustfa.org/
Trout information for both the consumer and industry. Information for the consumer includes: Tips and handling, farm-raised trout, trout tips, handling how-to's, prep pointers, and nutritional values. Industry information includes: Salmonid Magazine, Quality Assurance, membership info, and other internet resources, as well as a trout recipe book.

American Tilapia Association
http://ag.arizona.edu/azaqua/ata.html
Access to information about the fish, which is the fastest growing aquaculture crop in the United States and around the world. We are pleased to provide this information to those already producing tilapia for the food industry, for those interested in joining the industry and to potential customers and consumers of farm-raised tilapia.

Aquacultural Engineering Society
http://www.aesweb.org/
Newsletter - The latest quarterly newsletter of the AES is now on-line. Philosophy & Purpose - The official philosophy, purpose and operation of AES for your reference. Officers & Board - The current AES officers and board of directors. On-line Application - Use this electronic version of the member application to join AES!

Aquaculture Network Information Center (AquaNIC)
http://aquanic.org
An excellent clearinghouse of aquaculture information available on the internet. It also provides information on how to obtain aquaculture information that is not on the internet.

Aquatic Eco-Systems
http://www.aquaticeco.com
One of the world's largest aquaculture products suppliers. Useful link for finding equipment and prices.

Aquatic Network - Information Service for the Aquatic World
http://www.aquanet.com/
Subject areas covered include aquaculture, conservation, fisheries, marine science and oceanography, maritime heritage, ocean engineering, and seafood.

Cayuga Aqua Ventures, LLC
http://www.bee.cornell.edu/aqua
Publisher's website of this textbook. The site also has the software and descriptions that was in earlier versions of the text. Now provided as free-ware.

Cornell Aquaculture Resources & Short Course
http://www.bee.cornell.edu/outreach/aquaculture/short-course
This is the yearly course we put on each year, typically in mid July. See links on left side of page

FINS
http://www.actwin.com/fish/index.php
An archive of information about aquariums. It covers both freshwater and marine, tropical and temperate.

Holder Timmons Engineering, LLC
http://www.holdertimmons.com /
This is the author's site for his private engineering services company. It also shows the associates with the company. Listed here just for convenience to the reader.

National Fisheries Institute
http://www.aboutseafood.com/
National Fisheries Institute is the largest trade association serving the US seafood industry. NFI has always been a clearinghouse of business-related information. This website offers dozens of links to government statistics, seafood companies and other web resources. A members-only section adds promotional materials, HACCP information and the latest news on import alerts, legislative updates and more.

Natural Resources, Agricultural, and Engineering Service (NRAES)
http://www.nraes.org/

Sea Web
http://www.seaweb.org
To address the growing issue of salmon and other finfish farming in North America, in 1998 SeaWeb established an information clearinghouse on aquaculture issues. We focus our educational outreach efforts on salmon farming in the states of Maine and Washington and the provinces of New Brunswick, Nova Scotia, and British Columbia but collect and analyze information on various farmed species from around the world.

Seafood NIC
http://seafood.ucdavis.edu/
Hosted by the Sea Grant Extension Program at UC Davis, the Seafood Network Information Center, or Seafood NIC, is an on-line home to the HACCP Alliance. It offers page after page of seafood-safety information, as well as training materials, seminar schedules and the FDA's official fishery hazard and control guides. The site provides generic HACCP plans for scores of seafood products and processes.

USDA Regional Aquaculture Centers
There are four regional aquaculture centers that are sponsored by the USDA; they are:

Northeastern Regional Aquaculture Center
Dr. Reginal Harrell, Director
University of Maryland
2113 Animal Sciences Building
College Park, MD 20742-2317
Phone: (301) 405-6085;
Fax. (301) 314-9412
Email: nrac@umd.edu
Website: **http://www.nrac.umd.edu/**
Represents: Connecticut, Delaware, Maine, Maryland, Massachusetts, New Hampshire, New Jersey, New York, Pennsylvania, Rhode Island, Vermont, West Virginia, and the District of Columbia.

North Central Regional Aquaculture Center (NCRAC)

Dr. Ted Batterson, Director
Michigan State University
13 Natural Resources Building
East Lansing, MI 48824-1222
Phone: 517-353-1962; Fax: 517-353-7181
Email: batters2@msu.edu
Website: **www.ncrac.org**
Represents: Illinois, Indiana, Iowa, Kansas, Michigan, Missouri, Minnesota, Nebraska, North Dakota, Ohio, South Dakota, Wisconsin

Southern Regional Aquaculture Center (SRAC)
Dr. Craig S. Tucker, Director
Mississippi State University
127 Experiment Station Road
P.O. Box 197
Stoneville, MS 38776
Phone: 662-686-3285; Fax: 662-686-3320
Email: ctucker@drec.msstate.edu
Website: **http://www.msstate.edu/dept/srac**
Represents: Alabama, Arkansas, Florida, Georgia, Kentucky, Louisiana, Oklahoma, Mississippi, North Carolina, Puerto Rico, South Carolina, Tennessee, Texas, Virginia, Virgin Islands

Western Regional Aquaculture Center (WRAC)
Dr. Graham Young, Director
School of Fisheries
Box 355020
Seattle, WA 98195
Phone: 206-685-2479; Fax: 206-685-4674
E-mail: grahamy@u.washington.edu
Website: **http://www.fish.washington.edu/wrac**
Represents: Alaska, Arizona, California, Colorado, Idaho, Montana, Nevada, New Mexico, Oregon, Utah, Washington, Wyoming

Center for Tropical & Subtropical Aquaculture (CTSA)
Dr. Cheng-Sheng Lee, Executive Director
The Oceanic Institute
Makapuu Point
41-202 Kalanianaole Highway
Waimanalo, HI 96795-1820
Phone: 808-259-3107; Fax: 808-259-8395
Email: cslee@oceanicinstitute.org
Website: **http://www.ctsa.org**
Represents: American Samoa, Commonwealth of the Northern Mariana Islands, Federated States of Micronesia, Guam, Hawaii, Republic of Palau, Republic of the Marshall Islands

World Aquaculture Society (WAS)
http://www.was.org/
An international nonprofit society with over 4,000 members in 94 countries. Founded in 1970, , its primary focus is to improve communication and information exchange within the diverse global aquaculture community.

1.14 Summary and the Cornell Short Course

The authors have spent significant or all parts of their careers in aquaculture. Cornell has periodically hosted a one-week short course that hundreds of professionals have attended. The course was co-hosted with the Conservation Fund's Freshwater Institute (Shepherdstown WV) for several years. The course is now once again (2007 and forward) being sponsored by Cornell University. We will hold the course in various parts of the country and around the world on an as-needed basis along with the annual summer 1-week course that is held in July. The course is also offered in a Distance Learning format (see details for both the hands on and the distance versions at:

WWW.BEE.CORNELL.EDU/AQUA

The materials developed over the years for this short course are the primary materials being used in this book.

The RAS short course emphasizes practical "how to" applications and concludes with a series of spreadsheets and computer programs to make the many tedious calculations necessary to design and manage an RAS easier. However, the computer programs are much more useful to the user if the basic fundamentals are first understood. This book attempts to provide a solid foundation for understanding the basics and also includes the computer software in the Appendix. Example problems are given throughout the Chapters and these example problems (with solutions) can be used as a first try when using the software. Have fun and enjoy the book.

Past students of the Short Course can be seen at:

WWW.BEE.CORNELL.EDU/AQUA

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CHAPTER 2

WATER QUALITY

2.0 INTRODUCTION

The success of a commercial aquaculture enterprise depends on providing the optimum environment for rapid growth at the minimum cost of resources and capital. One of the major advantages of intensive recirculation systems is the ability to manage the aquatic environment and critical water quality parameters to optimize fish health and growth rates. Although the aquatic environment is a complex eco-system consisting of multiple water quality variables, it is fortunate that only a few of these parameters play decisive roles. These critical parameters are temperature, pH and concentrations of dissolved oxygen, ammonia, nitrite, CO_2, alkalinity and suspended solids. Each individual parameter is important, but it is the aggregate and interrelationship of all the parameters that influence the health and growth rate of the fish.

Each water quality parameter interacts with and influences other parameters, sometimes in complex ways. Concentrations of any one parameter that would be harmless in one situation can be toxic in another. For example, when aeration and degassing problems occur, carbon dioxide levels will generally become high, while at the same time dissolved oxygen levels become low. The result of this particular situation is that not only is there less oxygen available to the fish, the fish are less able to use the oxygen that is available. The high carbon dioxide level of the water affects the fishes' blood capacity to transport oxygen, aggravating the stress imposed by low dissolved oxygen levels. Another excellent example of the complex interaction among water quality parameters is the relationship between pH and the toxicity of ammonia. As will be discussed later, the unionized fraction of the total ammonia concentration is much more toxic than the ionized form (ammonium), and at low pH, most of the ammonia in the water is in the non-toxic ionized form. However, increasing the pH by only one unit, i.e., from 6.5 to 7.5, increases the concentration of the toxic unionized ammonia concentration by a factor of ten. Simply adding baking soda (or another base) to a system to increase its alkalinity can inadvertently increase the unionized ammonia to toxic levels. The result, for one of the authors, was 32 tanks of angelfish swimming upside down. This catastrophic

result provided a lesson learned the hard way about why it is important to understand the interrelationships of these parameters, and to routinely monitor as many critical water quality parameters as possible and when adjustments are needed, to make them carefully and slowly.

The relationship between water quality parameters and their effect on fish growth rate and health is complicated. For example, fish lack the means to control their body temperature and maintain it independent of the environment. Environmental temperature changes affect the fishes' rate of biochemical reactions, which leads to different metabolic and oxygen consumption rates. At the lower ranges of the species tolerable temperature range, these rates decrease. As water temperatures increase, fish become more active and consume more dissolved oxygen, while simultaneously producing more carbon dioxide and other excretory products, such as ammonia. These increasing rates of consumption of necessary elements and production of detrimental elements can have a direct effect on overall fish health and survival if these parameters are allowed to exceed nominal values. If not corrected, the fish will become stressed to some degree. Even low levels of stress can have adverse long-term consequences in the form of reduced growth rates or mortality due to opportunistic organisms that take advantage of the stressed fish.

2.1 PHYSICAL PROPERTIES

Several physical properties of water are important for the understanding of some of the engineering concepts that follow. Water has several very unique properties that make life on the planet possible. One of these is its density as a function of temperature, Table 2.1. Maximum density of pure water is reached at 3.98°C. A large discontinuity occurs at the freezing point, where the density drops drastically. Thus, ice is less dense than water and floats. If water did not exhibit this unusual density pattern associated with temperature, lakes and oceans would freeze from the bottom up and life, as we know it, would be impossible. The addition of salt and other impurities also increases the density of water. For example, seawater at 35 ppt salt has a density of 1.028 g/cm^3, as compared to 1.000 g/cm^3 for pure water. Thus, the density of water increases as salinity increases and/or temperature decreases. These density differences are the major driving force for overturn of freshwater systems and the vertical circulation patterns in the oceans.

Water viscosity is a measure of a fluid's resistance to shear, and is called absolute or dynamic viscosity. Highly viscous fluids, like

molasses or motor oil, flow very slowly when subject to a shear stress. Absolute viscosity is measured in centipoises, and water at 20°C has a value of 1.0 centipoise, compared to air, which has a value of 0.17 centipoise, at this temperature. The absolute viscosity divided by the liquid density is the given the name kinematic viscosity. Table 2.1 shows the variation of density, absolute viscosity, and kinematic viscosity of water at a range of temperatures. Viscosity increases as temperature decreases. Thus, pumping costs will increase slightly as water temperature decreases because viscosity and density both increase.

Vapor pressure is the pressure exerted by the gaseous phase of a material when in equilibrium with its solid or liquid phase. Another way of expressing vapor pressure is the pressure at which a liquid just begins to boil and change to a vapor (Lawson, 1995). Vapor pressure of pure water is a function of temperature and increases with an increase in temperature, Table 2.1. The addition of salt to water lowers its vapor pressure. In a closed pipe, water may change phase from a liquid to a vapor because of a reduction in pressure, even though the temperature remains unchanged. This can occur on the suction side of pipe, where vapor bubbles form in local regions of very low pressure and then collapse when they move into regions of higher pressure downstream. This process is called cavitation. When a pump is cavitating, the collapsing bubbles contain a great deal of energy and can cause considerable damage to pump impellers and pipes.

Table 2.1 Physical Properties of Water

Temp. °C	Density kg/m^3	Kinematic Viscosity (m^2/s)·E-06	Vapor Pressure mm Hg
0	999.84	1.79	4.8
1	999.90	1.73	5.1
2	999.94	1.68	5.4
3	999.97	1.62	5.8
4	1000.00	1.57	6.2
5	999.97	1.52	6.6
10	999.70	1.31	9.1
15	999.10	1.13	12.5
20	998.21	0.99	17.3
25	997.05	0.88	23.9
30	995.65	0.80	33.0
35	994.04	0.72	45.5
40	992.22	0.66	62.8

Formulae for generating water physical properties (T is in °C):

$$\textbf{Density}\ (kg/m^3) = 999.842594+6.793952E\text{-}2\cdot T -9.095290E\text{-}3\cdot T^2+1.001685E\text{-}4\cdot T^3 - 1.120083E\text{-}6\cdot T^4+6.536336E\text{-}9\cdot T^5 \quad (2.1)$$

$$\textbf{Vapor Pressure}\ (mm\ Hg) = 4.7603\ e^{0.0645T} \quad (2.2)$$

$$\textbf{Kinematic Viscosity}\ (m^2/s\ 10^{-6}) = -9.9653E\text{-}6\cdot T^3 +0.001143T^2-0.05807T+1.7851 \quad (2.3)$$

2.2 WATER QUANTITY REQUIREMENTS

During the initial site selection process, one of the most critical factors to consider is the availability of an adequate water supply for both the initial facility and any planned (or imagined) expansion. When it comes to the availability of water, too much is definitely better than too little. After all, one is constructing an **AQUA**culture facility. The amount of water needed will depend on several factors such as species, density, management practices, production technology, and the degree of risk one is willing to accept. At a minimum, sufficient quantities of water are needed to routinely fill production tanks within a reasonable time (24-48 hrs), provide for routine and emergency flushing of tanks, filter backwashing, facility wash down and clean-up, and domestic requirements. A good rule of thumb is to have sufficient water available to provide a 100% water exchange of total system volume per day (Timmons, 2000). Thus for a total system volume of 379 m^3 (100,000 gallons), a water supply is required that is able to provide 379 m^3 per day or 0.26 m^3/min (70 gpm). This may be reduced some for a warm water species.

Beyond the minimum exchange volume, the additional quantity of new water required for any given system is directly dependent on the degree of reuse or recycling of the "old" water that is already available in the system. There are numerous reasons to reuse the supply water as much as possible, besides the obvious decrease in water demand and reduced discharge water that must be treated. These include the reduction of water heating or cooling requirements, which are major factors in the economics of many warm-water species production systems. Additionally, and becoming increasingly important, recycling of system waters results in the reduction in wastewater discharge and the corresponding costs involved in the treatment of large discharge flow streams.

There are three categories of reuse systems: serial-reuse systems, partial-reuse systems, and fully recirculating systems. Partial-reuse systems reuse a greater percentage of total system volume than do serial-reuse systems, and the fully recirculating systems reuse a greater percentage of total system volume than do the partial-reuse systems. The degree of water reuse affects the depletion/accumulation rate of important water quality parameters. The greater the reuse rate, the more conditioning the reused water must have in order to restore the water quality to target parameters. Typically, the most important and first encountered limiting factor that governs the density of fish reared in a system is the concentration of dissolved oxygen. The next most important factors are the amounts of unionized ammonia and dissolved carbon dioxide levels. These two parameters are interconnected. This is due to the direct effect that dissolved carbon dioxide has on pH and the relationship of pH to the toxicity of ammonia-nitrogen. As the dissolved carbon dioxide levels decrease, the pH increases, which in turn increases the toxicity of the total ammonia-nitrogen in the system. For example, if salmonids are the species being grown, the maximum upper safe limit for chronic exposure to carbon dioxide is from <9 to 30 mg/L, and for unionized ammonia-nitrogen <0.0125 to 0.03 mg/L. The chosen reuse system must be able to sustain the necessary levels of dissolved oxygen while also keeping the amounts of dissolved carbon dioxide, unionized ammonia, and the pH below their respective limiting levels.

Serial-reuse systems have been used extensively in trout and salmonid raceway production systems, where the limiting water quality factor is usually the dissolved oxygen concentration between raceway sections. By adding oxygen, the water can be used over again in the next section of the raceway, until ultimately the accumulated ammonia levels become too high. This simple concept has significantly increased raceway production, but at a higher system cost due to the requirements for more sophisticated oxygenation systems and monitoring systems. Economic risk is also higher, because there are more fish in the raceway that are subject to a catastrophic event.

Partial-reuse systems are an alternative culture system that can sustain high production densities on less than 20% of the total flow that would be required to grow the same quantity of fish in a serial-reuse system (Summerfelt, et al., 2000). Some partial-reuse systems separate the solid wastes from the main water recirculation loop by using a dual-drain system. In this system, the circular production tank is used as a "swirl" separator to concentrate the solids into the center drain. From

there, they are removed with a relatively small discharge flow stream (15–20% of the quantity of water in the tank per day). The majority of the water flow leaving the tank, i.e., the remaining 80–85%, is discharged from the tank through a fish-excluding port located approximately midway on the tank sidewall. This flow stream is relatively free of settleable solids and can be easily treated with a high-capacity microscreen filter, carbon dioxide stripping system, and an oxygenation system. The ammonia levels are controlled by dilution with make-up water, typically 10–20% of the flow (a volume approximately equal to the solids waste discharge flow from the center drain), and by controlling the system pH. The key operating water quality parameter in this type of system is the amount of dissolved carbon dioxide. Adjusting the amount of carbon dioxide stripped by the degassing system will control the system's pH. When the system is operated so that the dissolved carbon dioxide level is the limiting water quality parameter, the water will have a low pH, and thus the corresponding maximum total ammonia-nitrogen level will also be well below the level at which it would become critical.

The nomograph in Fig. 2.1 provides an estimate of the maximum total ammonia-nitrogen that can be allowed to accumulate in a partial-reuse system, depending upon the water's alkalinity and assumed maximum limits on the concentrations of dissolved carbon dioxide and unionized ammonia (Summerfelt et al. 2000). Thus setting the maximum limits on the concentrations of dissolved carbon dioxide and unionized ammonia, at a given alkalinity, will set the minimum operating pH and the maximum allowable total ammonia-nitrogen.

2.3 WATER SOURCES

One of the most important requirements for a successful aquaculture facility site is a high quality water source, with sufficient capacity to provide for initial needs and contemplated future expansions. Thus, when choosing an aquaculture site, the water supply must be thoroughly investigated and quantified in terms of both quality and quantity. Aquaculture is unlike other agriculture activities, in that it requires a continuous supply of high quality water. Most wells operate only periodically, so a traditional well pump test is usually conducted for only a short time, to measure the well's ability to deliver a certain quantity of water within a certain time. However, under normal aquaculture operations, the well is pumped continuously. The result can be that the actual productivity of the well is significantly less than indicated by the short duration test. This will result in serious difficulties down the road.

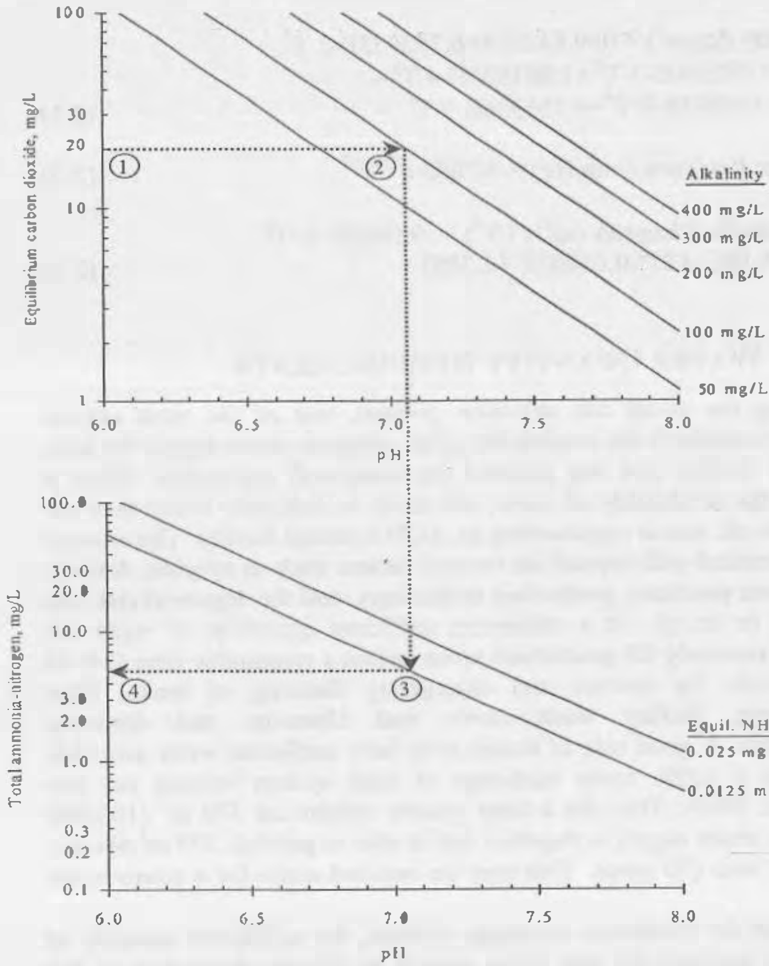


Figure 2.1 Dependence of dissolved carbon dioxide concentrations on pH and alkalinity (top graph), and unionized ammonia concentrations and total ammonia-nitrogen (bottom graph), according to acid-base equilibrium at 15°C. Dashed lines indicate how setting a maximum limit of 20 mg/L on the concentration of dissolved carbon dioxide (1) and of 0.0125 mg/L on unionized ammonia nitrogen (3), at an alkalinity of 100 mg/L (2), will set the minimum operating pH (about 7.06) and the maximum allowable TAN (4) (about 5.0 mg/L), from Summerfelt et al. 2000.

There are several sources of water for aquaculture operations, each with distinct advantages and disadvantages. Since the whole point of a recirculating aquaculture system (RAS) is to minimize water usage, the two most often used water sources are groundwater and municipal water supplies. Both normally have the quality, quantity, and reliability required, so the actual choice between them is based on availability and economics. Surface waters are generally not used, due to the higher risks of contamination by pollutants, fish eggs, insect larva, disease microorganisms, and wide seasonal temperature variations.

One of the major advantages of groundwater sources is their constant temperature throughout the year. However, shallow sources of groundwater approximate the mean air temperature of the area. The chemistry of groundwater is directly dependent on the geology of the area surrounding the source. In limestone areas, groundwater is bard, and high in calcium and inorganic carbon. In areas of granite formation, the groundwater tends to be soft, low in dissolved minerals and inorganic carbon. As will be discussed later, there are advantages and disadvantages to both, emphasizing the need for early extensive water quality testing.

The disadvantages of groundwater, especially from deep wells, are the high concentrations of dissolved toxic gases such as hydrogen sulfide (H_2S), methane (CH_4), and carbon dioxide (CO_2). Most groundwater contains little or no dissolved oxygen, because of the biological processes occurring in the aquifer recharge zone. In addition, once pumped to the surface, groundwater often is supersaturated with nitrogen gas, argon, and carbon dioxide. Thus, the groundwater must be initially conditioned by some form of aeration and degassing to remove the excess gases and add oxygen. High concentrations of dissolved iron can also be a problem. Once aerated, the iron precipitates out of solution as iron oxide and is often removed by some form of granular filtration or settling basin.

Another potential source of water is from a municipal water supply. However, it must be kept in mind that these sources are designed and treated to safeguard the health of humans, i.e., chemicals such as chlorine (1.0 mg/L) and fluorine are added. To be useable in RAS, these chemicals must be neutralized either chemically (sodium thiosulfate to remove chlorine and chloramines) or by filtration (activated carbon). Chlorine is very detrimental to fish and levels as low as 0.02 mg/L are stressful.

2.4 WATER QUALITY STANDARDS

The very nature of aquaculture makes it almost impossible to formulate a definitive "one size fits all" list of water quality standards. The wide range of species, temperature regimes, and production techniques makes any such list only “recommendations”, at best. In intensive recirculation systems, the species of choice is raised in a kind of chemical soup consisting of numerous physical, chemical, and biological factors that are interrelated in a complex series of physico-bio-chemical reactions. These reactions affect every aspect of the culture from fish survival and growth rates to biofilter performance and solids removal. A basic understanding of water chemistry is critical for the success of any intensive system.

Species handbooks, fish hatchery manuals, or aquaculture books are an excellent source of information for water quality information and current culture protocols. Some of the most helpful references for species commonly reared in the Americas are presented below (Colt, 2006):

Genus, species, or group	Reference
Salmonids	Barton (1996), Piper et al. (1982), Sigma (1983), Wedemeyer (1996)
Temperate basses	Tomasso (1997)
Channel catfish	Boyd and Tucker (1998), Stickney (1993)
Black basses	Williamson et al. (1993)
Sunfishes	Williamson et al. (1993)
Walleye	Nickum and Stickney (1993)
Pikes	Westers and Stickney (1993)
Tilapia	Costa-Pierce and Rakocy (1997)
Sturgeon	Conte (1988)
Marine fish	Poxton and Allouse (1982)
Marine larval fishes	Brownell (1980a,b)
General	Wickins (1981)

While these sources are very useful, it is important to realize that in general, they are based on standard toxicity tests, usually short time exposure to constant concentrations. Commonly, much of information contained in these sources is based on the experiences and practices of state, tribal, and federal hatchery programs (Colt, 2006). Current information on acceptable water quality parameters from large scale commercial enterprises is very difficult to find.

Over the years, standardized toxicity testing protocols for aquatic animals have been developed. Most of these toxicity studies are conducted on juvenile animals for a relatively short time. The animals

are exposed to constant concentrations of toxic substances, while maintaining other water quality parameters within an acceptable range. The extrapolation of these types of experiments to the entire production cycle and to combinations of environmental parameters (for example, high total ammonia, and carbon dioxide in combination with supersaturated dissolved oxygen) is difficult if not impossible (Colt, 2006). These types of exposures seldom occur in real systems. A real world evaluation of ammonia toxicity might test the impact of diel varying ammonia concentration on the growth of 300–400 g tilapia under high carbon dioxide levels when DO is maintained at 70% of saturation, and alkalinity is low.

The water quality criteria for the design of culture systems depend strongly on the objectives of the project, species and life stage grown. The impact of water quality on growth is probably the most important consideration. Other impacts include fin quality and appearance which is important for species sold in the 'live-market'; tissue quality such as texture, taste and odor and finally trace contaminates in the tissue such as methymercury, PCT, DDT or dioxins (Colt, 2006). Development of species or system specific water quality criteria is time consuming and expensive (Colt, 2006). A summary of important water quality parameters is presented in Table 2.2 and provides very general recommendations of water quality criteria for each parameter. Colt (2006) provides additional details, especially regarding nitrite concentrations.

Several other water quality parameters are currently being carefully reviewed due to the uncertainty as to their impact on production. The effect of high levels of fine solids and organic compounds is unknown at this time, but has been implicated in disease outbreaks (Bullock, et al., 1994). The leaching of surface-active compounds from uneaten feed and fecal matter and the resulting depression of surface tension may be an important design consideration (Colt, 2006). In limited exchange systems, there is a potential to build-up toxic concentrations of some heavy metals, such as cadmium, copper and zinc from the corrosion of pipes and fittings and from vitamin premix. The toxicity of heavy metals depends strongly on water chemistry. Finally, there is limited data on the impact of pheromones, endocrine-disrupting chemicals (EDCs), and chemicals leached from paints, PVC components, and liners. These chemicals can exert profound and adverse effects on aquatic animals by interfering with the endocrine systems and potentially resulting in reduced fertility (Colt, 2006) and lower growth rates. The environmental chemistry of different EDCs is complex and the analysis is costly.

Table 2.2a Water Quality Criteria for Aquaculture

Parameter	Concentration (mg/L)
Alkalinity (as $CaCO_3$)	50–300
Aluminum (Al)	<0.01
Ammonia (NH_3-N unionized)	<0.0125 (Salmonids)
Ammonia (TAN) Cool-water fish	<1.0
Ammonia (TAN) Warm-water fish	<3.0
Arsenic (As)	<0.05
Barium (Ba)	<5
Calcium (Ca)	4–160
Carbon Dioxide (CO_2)	
Tolerant Species (tilapia)	<60
Sensitive Species (salmonids)	<20
Chlorine (Cl)	<0.003
Hardness, Total (as $CaCO_3$)	>100
Hydrogen cyanide (HCN)	<0.005
Hydrogen sulfide (H_2S)	<0.002
Iron (Fe)	<0.15
Lead (Pb)	<0.02
Magnesium (Mg)	<15
Manganese (Mn)	<0.01
Mercury (Hg)	<0.02
Nitrogen (N_2)	<110% total gas pressure <103% as nitrogen gas
Nitrite (NO_2)	<1, 0.1 in soft water
Nitrate (NO_3)	0–400 or higher
Nickel (Ni)	<0.1
Oxygen Dissolved (DO)	>5
(see Chapter 4 for more detail)	> 90 mm Hg partial pressure
Ozone (O_3)	<0.005
PCBs	<0.002
pH	6.5–8.5
Phosphorous (P)	0.01–3.0
Potassium (K)	<5
Salinity	depends on salt or fresh species
Selenium (Se)	<0.01
Silver (Ag)	<0.003
Sodium (Na)	<75
Sulfate (SO_4)	<50
TGP (total gas pressure)	<105% (species dependent)
Sulfur (S)	<1
Total dissolved solids (TDS)	<400 (site specific and species specific; use as rough guideline)
Total suspended solids (TSS)	10 to 80 (species dependent)
Uranium (U)	<0.1
Vanadium (V)	<0.1

Source: Meade, 1985; Piper et al. 1982; Lawson, 1995

Table 2.2b Heavy Metal Water Quality Criteria

Metal (µg/L)	Freshwater 500*	100*	10*	1*	Seawater
Copper	35	9	1.3	0.18	3.1
Zinc	460	120	17	2.4	81
Cadmium	0.75	0.25	0.049	0.01	8.8

*Hardness (mg/L as $CaCO_3$)
Source: Colt, 2006

2.5 Water Quality Parameters

Dissolved Oxygen

Of all the water quality parameters, dissolved oxygen is the most important and most critical parameter, requiring continuous monitoring in intensive production systems. Nature played a cruel joke on aquaculture when she decided that the saturation concentration of dissolved oxygen would be higher at low temperature and lower at high temperatures. This condition is exactly the opposite of what fish require for basic metabolism and food conversion, which is higher at high temperatures and lower at low temperatures. Although the air we breathe contains 21% oxygen, oxygen is only slightly soluble in water. As a result, aquatic species must spend a great deal of energy to remove the dissolved oxygen from water, as compared to the energy that land dwelling species expend to obtain oxygen from the air. In addition, the solubility of oxygen decreases as salinity increase. Both barometric pressure and altitude also directly affect oxygen concentration. Table 2.3 lists the saturation oxygen concentration in water as a function of salinity and temperature.

It is difficult to specify critical dissolved oxygen concentrations, because the response to low dissolved oxygen is not life-or-death, but a continuum of physiological effects. In addition, these effects are influenced by the exposure time, the size, and health of the fish, water temperature, concentration of carbon dioxide, and other environmental conditions. In general, though, warm water fish feed best, grow fastest, and are healthiest when dissolved oxygen concentrations are above about 5 mg/L. However, concentrations of dissolved oxygen greater than this level of saturation appear to provide no additional benefit to fish. The gills can only transfer so much oxygen to the blood, and are very near or at the maximum transfer capability when the environmental dissolved oxygen concentrations are at the recommended concentrations. Higher oxygen concentrations in the water do not result in any additional oxygen carried by the blood stream.

Table 2.3 Dissolved Oxygen (mg O_2 per Liter, ppm) at Saturation in Freshwater, Brackish Water, and Seawater at Different Temperatures

Temp (°C)	Salinity, parts per thousand 0	5	10	15	20	25	30	35	40
0	14.602	14.112	13.638	13.180	12.737	12.309	11.896	11.497	11.111
1	14.198	13.725	13.268	12.825	12.398	11.984	11.585	11.198	10.825
2	13.813	13.356	12.914	12.487	12.073	11.674	11.287	10.913	10.552
3	13.445	13.004	12.576	12.163	11.763	11.376	11.003	10.641	10.291
4	13.094	12.667	12.253	11.853	11.467	11.092	10.730	10.380	10.042
5	12.757	12.344	11.944	11.557	11.183	10.820	10.470	10.131	9.802
6	12.436	12.036	11.648	11.274	10.911	10.560	10.220	9.892	9.573
7	12.127	11.740	11.365	11.002	10.651	10.311	9.981	9.662	9.354
8	11.832	11.457	11.093	10.742	10.401	10.071	9.752	9.443	9.143
9	11.549	11.185	10.833	10.492	10.162	9.842	9.532	9.232	8.941
10	11.277	10.925	10.583	10.252	9.932	9.621	9.321	9.029	8.747
11	11.016	10.674	10.343	10.022	9.711	9.410	9.118	8.835	8.561
12	10.766	10.434	10.113	9.80.1	9.499	9.207	8.923	8.648	8.381
13	10.525	10.203	9.891	9.589	9.295	9.011	8.735	8.468	8.209
14	10.294	9.981	9.678	9.384	9.099	8.823	8.555	8.295	8.043
15	10.072	9.768	9.473	9.188	8.911	8.642	8.381	8.129	7.883
16	9.858	9.562	9.276	8.998	8.729	8.468	8.214	7.968	7.730
17	9.651	9.364	9.086	8.816	8.554	8.300	8.053	7.814	7.581
18	9.453	9.174	8.903	8.640	8.385	8.138	7.898	7.664	7.438
19	9.265	8.990	8.726	8.471	2.222	7.982	7.748	7.521	7.300
20	9.077	8.812	8.556	8.307	8.065	7.831	7.603	7.382	7.167
21	8.898	8.641	8.392	8.149	7.914	7.685	7.463	7.248	7.038
22	8.726	8.476	8.233	7.997	7.767	7.545	7.328	7.118	6.914
23	8.560	8.316	8.080	7.849	7.626	7.409	7.198	6.993	6.794
24	8.400	8.162	7.931	7.707	7.489	7.277	7.072	6.872	6.677
25	8.244	8.013	7.788	7.569	7.357	7.150	6.950	6.754	6.565
26	8.094	7.868	7.649	7.436	7.229	7.027	6.831	6.641	6.456
27	7.949	7.729	7.515	7.307	7.105	6.908	6.717	6.531	6.350
28	7.808	7.593	7.385	7.182	6.984	6.792	6.606	6.424	6.248
29	7.671	7.462	7.259	7.060	6.868	6.680	6.498	6.321	6.148
30	7.539	7.335	7.136	6.943	6.755	6.572	6.394	6.221	6.052
31	7.411	7.212	7.018	6.829	6.645	6.466	6.293	6.123	5.959
32	7.287	7.092	6.903	6.718	6.539	6.364	6.194	6.029	5.868
33	7.166	6.976	6.791	6.611	6.435	6.265	6.099	5.937	5.779
34	7.049	6.863	6.682	6.506	6.335	6.168	6.006	5.848	5.694
35	6.935	6.753	6.577	6.405	6.237	6.074	5.915	5.761	5.610
36	6.824	6.647	6.474	6.306	6.142	5.983	5.828	5.676	5.529
37	6.716	6.543	6.374	6.210	6.050	5.894	5.742	5.594	5.450
38	6.612	6.442	6.277	6.117	5.960	5.807	5.659	5.514	5.373
39	6.509	6.344	6.183	6.025	5.872	5.723	5.577	5.436	5.297
40	6.410	6.248	6.091	5.937	5.787	5.641	5.498	5.360	5.224

For salmonids, as a group, the rearing unit effluent should contain from 6.0 to 8.0 mg/L dissolved oxygen (DO). For catfish and tilapia, allowable minimum levels are much lower than for salmonids, e.g., 2 or 3 mg/L, while it is certainly recommended to stay much closer to 5 or 6 mg/L. This variation in what the allowable minimums are has to do with the fact that partial oxygen pressure (pO_2) appears to be a more valid way to determine the lower limits. A pO_2 of 90 mm Hg seems to be a reasonable target for salmonids (Downey and Klontz, 1981). The atmosphere contains 21% oxygen, and at standard pressure of 760 mm Hg at 20°C, this represents a pO_2 of 0.21 • (760–17.54)[a] or 155.9 mm Hg. and corresponds to a dissolved oxygen saturation of 9.1 mg/L. Therefore, 90 mm Hg corresponds to 5.2 mg/L DO. If the temperature is 5°C, pO_2 corresponds to 0.21 • (760 – 6.54) or 158.2 mm Hg and corresponds to a DO saturation 12.8 mg/L. In this case, 90 mm Hg corresponds to 7.3 mg/L. As noticed, the warmer the water the lower the effluent DO in mg/L can go, while still representing a pO_2 of 90 mm Hg. Applying 90 mm pO_2 @ 30°C for tilapia would set a target value of 4.4 mg/L for culture tank oxygen levels.

Temperature

Water temperature is second only to dissolved oxygen in importance and impact on the economic viability of a commercial aquaculture venture. Temperature directly affects the physiological processes, such as respiration rate, efficiency of feeding and assimilation, growth, behavior, and reproduction. Fish have traditionally been grouped into three classifications depending on their temperature preference: cold-water, cool-water, and warm-water. Cold-water species prefer water temperatures under 15°C (60°F), cool-water species between 15°–20°C (60°–68°F), and warm-water species above 20°C (68°F). These are not exact definitions and several factors are involved in determining the tolerance of fish to different temperatures; these include species, age, size, and past thermal history.

Fish are classified as poikilothermic or cold-blooded, which means that their body temperature is approximately the same as their surrounding environment. Therefore, each species has an optimum temperature range that maximizes growth and an upper and lower limit beyond which they cannot survive. Within the species tolerable temperature range, growth rates increase as the water temperature increases, until the optimum temperature is reached. Above the optimum temperature, the increased energy requirements for food conversion and

[a] 17.54 mm Hg is the vapor pressure of water at 20°C

other metabolic processes ensure that the law of diminishing returns applies. In addition, at higher than optimum temperatures, the fish food conversion ratios are lower. Further temperature increases beyond optimum are of no benefit, and may in fact approach lethal levels. Thus to insure maximum growth and minimize stress, system temperatures need to be maintained as close as possible to optimum value. Optimum temperature range for several representative species is presented in Table 2.4.

Table 2.4 Optimum Temperature Ranges (°C) for Representative Aquaculture Species

Species	Temp. Range	Source
brown trout (*Salmo trutta*)	12–14°	Aston, 1981
rainbow trout (*Oncorhynchus mykiss*)	14–16°	Aston, 1981
brook trout (*Salvelinus fontinalus*)	7–13°	Piper, et al. 1982
Chinook salmon (*Oncorhynchus tshawytscha*)	10–14°	Piper, et al. 1982
Coho salmon (*Oncorhynchus kisutch*)	9–14°	Piper, et al. 1982
Atlantic salmon (*Salmo salar*)	15°	Aston, 1981
sockeye salmon (*Oncorhynchus nerda*)	15°	Aston, 1981
turbot (*Scophthalmus maximus*)	19°	Aston, 1981
sole (*Solea solea*)	15°	Aston, 1981
channel catfish (*Ictalurus punctatus*)	25–30°	Tucker & Robinson, 1990
striped bass (*Moronoe saxatilis*)	13–24°	Piper, et al. 1982
red drum (*Sciaenops ocellatus*)	22–28°	Piper, et al. 1982
largemouth bass (*Micropterus salmoides*)	25–30°	Piper, et al. 1982
European eel (*Anguilla anguilla*)	22–26°	Aston, 1981
Japanese eel (*A. jabonica*)	24–28°	Aston, 1981
carp (*Cypriums carpio*)	25–30°	Aston, 1981
mullet (*Mugil cephalus*)	28°	Aston, 1981
tilapia (*Sarotheorodon. sp.*)	28–32°	Aston, 1981

Ammonia / Nitrite / Nitrate

Nitrogen is an essential nutrient for all living organisms, and is found in proteins, nucleic acids, adenosine phosphates, pyridine nucleotides, and pigments. However, nitrogen is required in relatively small quantities, and physiological needs are easily satisfied. Excess quantities then become nitrogenous wastes, and removal is necessary. The fish create and expel various nitrogenous waste products through gill diffusion, gill

cation exchange, and urine and feces excretion. In addition to the ammonia, urea, uric acid, and amino acid excreted by the fish, nitrogenous wastes accumulate from the organic debris of dead and dying organisms, uneaten feed, and from nitrogen gas in the atmosphere. Decomposing these nitrogenous compounds is particularly important in intensive RAS because of the toxicity of ammonia, nitrite, and to some extent, nitrate.

Ammonia, nitrite, and nitrate are all highly soluble in water. Ammonia exists in two forms: unionized NH_3, and ionized NH_4^+ (ammonium). The relative concentration of the two forms of ammonia is primarily a function of water pH, salinity, and temperature. The sum of the two ($NH_4^+ + NH_3$) is called total ammonia nitrogen or simply ammonia. It is common in chemistry to express inorganic nitrogen compounds in terms of the nitrogen they contain, i.e., NH_4^+–N (ionized ammonia nitrogen), NH_3–N (unionized ammonia nitrogen), NO_2^-–N (nitrite nitrogen) and NO_3^-–N (nitrate nitrogen). This allows for easier computation of total ammonia-nitrogen (TAN = NH_4^+–N + NH_3–N) and easy conversion between the various stages of nitrification.

DEFINITIONS

- NH_3–N = unionized ammonia nitrogen (ammonia)
- NH_4^+–N = ionized ammonia nitrogen (ammonium)
- TAN = NH_4^+–N + NH_3–N
- NO_2^-–N = nitrite nitrogen
- NO_3^-–N = nitrate nitrogen

Unionized ammonia (NH_3–N) is the most toxic form of ammonia because of its ability to move across cell membranes, so the toxicity of TAN is dependent on the percentage present in the unionized form. An increase in pH, temperature, or salinity increases the proportion of the unionized form of ammonia nitrogen. The concentration of NH_3-N can be computed knowing the pH from the following equation:

$$NH_3 - N = \frac{TAN}{\left(1 + 10^{(pKa - pH)}\right)} \qquad (2.4)$$

where TAN is the measured concentration of total ammonia nitrogen (mg/L); pK_a, the acidity constant for the reaction (9.40 in fresh water at 20 °C); pH, the measured pH of the solution; NH_3- N, computed concentration in (mg/L).

For example, given a TAN = 5.0 mg/L in fresh water at 20°C and a pH of 7.0, the concentration of unionized ammonia is only 0.020 mg/L, which has a negligible impact on most fish. However, at a pH of 9.0, the unionized ammonia increases to 1.43 mg/L, killing most species of fish in hours. The fraction of unionized ammonia at different temperatures and pH are included in the Appendix.

Ammonia appears to have a direct effect on the growth of aquatic animals. Unionized ammonia is toxic to fish at low concentrations, with 96-hour LC_{50}'s varying widely by species from as low as 0.08 mg/L NH_3-N for pink salmon to 2.2 mg/L NH_3-N for common carp. In general, warm-water fish are more tolerant to ammonia toxicity than cold-water fish, and freshwater fish are more tolerant than saltwater fish. In general, for commercial production, unionized ammonia concentrations should be held below 0.05 mg/L and TAN concentrations below 1.0 mg/L for long-term exposure. Table 2.5 summarizes the effects of temperature and pH on the percentage of free ammonia in freshwater.

Table 2.5 Percentage of Free Ammonia (as NH3) in Freshwater at Varying pH and Water Temperature, (Lawson,1995)

pH	10°C (50°F)	15°C (59°F)	20°C (68°F)	25°C (77°F)	30°C (86°F)
6.0	---	--	--	0.1	0.1
6.5	0.1	0.1	0.1	0.2	0.3
7.0	0.2	0.3	0.4	0.6	0.8
7.5	0.6	0.9	1.2	1.8	2.5
8.0	1.8	2.7	3.8	5.4	7.5
8.5	5.6	8.0	11.2	15.3	20.3
9.0	15.7	21.5	28.4	36.3	44.6
9.5	37.1	46.4	55.7	64.3	71.8
10.0	65.1	73.3	79.9	85.1	89.0

Nitrite is the ionized form of the relatively strong acid, nitrous acid and is the intermediate product in the process of nitrification of ammonia to nitrate. Although nitrite is converted to nitrate relatively quickly by ozone or by the nitrifying bacteria in a properly balanced biofilter, it is a problem in recirculating systems because it can build up easily if the second bacterial driven stage of the nitrification process is not working properly. Therefore, it is an important water quality parameter to monitor and correct if acceptable limits are exceeded. Nitrite is toxic because it affects the blood hemoglobin's ability to carry oxygen. When nitrite enters the bloodstream, it oxidizes the iron in the hemoglobin molecule

from the ferrous state to the ferric state. The resulting product is called methemoglobin, which has a characteristic brown color, hence the common name "brown-blood disease". The amount of nitrite entering the blood depends on the ratio of nitrite to chloride in the water, in that increased levels of chloride reduce the amount of nitrite absorption. Chloride levels can be increased by adding ordinary salt (sodium chloride) or calcium chloride. At least a 20:1 ratio of chloride to nitrite-nitrogen (Cl: NO_2–N) is recommended for channel catfish in ponds, tilapia, and rainbow trout.

Nitrate is the end-product of nitrification and is the least toxic of the nitrogen compounds, with a 96-h LC_{50} value usually exceeding 1000 mg NO_3-N /L. In recirculation systems, nitrate levels are usually controlled by daily water exchanges. In systems with low water exchange or high hydraulic retention times, denitrification has become increasingly important (see Chapter 9).

PH

The pH value expresses the intensity of the acidic or basic characteristic of water. In simplified chemical terms, pH is the negative logarithm of the hydrogen ion concentration. The pH scale ranges from 0 to 14, with a pH of 7.0 corresponding to the neutral point. Values of pH below 7.0 are acidic (the H^+ ion predominates), and above 7.0, values are basic or alkaline (the OH^+ ion predominates). The pH of most groundwater's and surface waters is buffered by the bicarbonate-carbonate system and has pH values between 5 and 9. Exceptions to this are groundwater with high concentrations of dissolved carbon dioxide, and water draining from some mines (acid mine drainage, AMD). AMD is the result of the oxidation of mineral sulfides, which becomes sulfuric acid. Seawater is buffered by the bicarbonate-borate system and has a relatively stable pH between 8.0 and 8.5. The optimum pH for the growth and health of most freshwater aquatic animals is in the range of 6.5 to 9.0.

Exposure to extreme pH can be stressful or lethal, but it is the indirect effects resulting from the interactions of pH with other variables that are more important in aquaculture. pH controls a wide variety of solubility and equilibria reactions, the most important of which is the relationship between the unionized and the ionized form of ammonia and nitrite. pH also affects the toxicity of hydrogen sulfide and metals such as copper, cadmium, zinc and aluminum.

ALKALINITY/HARDNESS

In broad terms, alkalinity is a measure of the pH-buffering capacity or the acid-neutralizing capacity of water. In chemical terms, alkalinity is defined as the total amount of titratable bases in water expressed as mg/L equivalent calcium carbonate ($CaCO_3$). Sometimes alkalinity is expressed as milliequivalents/liter, where 1 meq/L equals 50 mg/L as $CaCO_3$. The principle ions that contribute to alkalinity are carbonate (CO_3^-) and bicarbonate (HCO_3^-). Table 2.6 provides a list of commonly used alkalinity supplements, their relative solubility, and their equivalents basis (see also Chapter 7 for discussion of nitrification and alkalinity consumption). See the appendix for hardness conversions to a variety of units.

Table 2.6 Alkalinity Supplement Properties (Bisogni and Timmons, 1994)

Chemical Formula	Common Name(s)	Equivalent Wt. (gm/eq.)	Solubility	Rate of solubilization
NaOH	sodium hydroxide	40	high	rapid
Na_2CO_3	sodium carbonate soda ash	53	high	rapid
$NaHCO_3$	sodium bicarbonate baking soda	83	high	rapid
$CaCO_3$	calcium carbonate calcite	50	moderate	moderate
CaO	slaked lime	28	high	moderate
$Ca(OH)_2$	calcium hydroxide hydrated lime	37	high	moderate
$CaMg(CO_3)_2$	dolomite	46	moderate	slow
$MgCO_3$	magnesium carbonate magnesite	42	moderate	slow
$Mg(OH)_2$	magnesium hydroxide brucite	29	moderate	slow

Note: Na compounds are highly soluble in water while Mg compounds have poor solubility. Ca compounds are intermediate. Mg compounds tend to dissolve very slowly, so may have application in situations requiring a long-term application. Na compounds may prove to be the most expensive.

Based on 100% pure compound. To calculate for impurities, divide the tabulated value by the pure fraction ({100%-impurities %}/100 gives pure fraction) to get the true value.

In practical terms, alkalinity is measured by titration with sulfuric or hydrochloric acid to the methyl orange endpoint (pH of 4.5). Alkalinity of freshwater ranges from less than 5 mg/L in soft water to over 500 mg/L, and is determined by the geology of the aquifer or watershed. The alkalinity of seawater is about 120 mg/L $CaCO_3$. Required alkalinity concentrations are directly linked to system pH and carbon dioxide concentrations. Maintaining carbon dioxide concentrations at less than 15 mg/L and pH between 7.0 and 7.4 requires an alkalinity concentration less than 70 (high pH condition) to 190 mg/L $CaCO_3$ (low pH condition). The relationship between pH, alkalinity, and CO_2 concentrations is shown in Fig. 2.2.

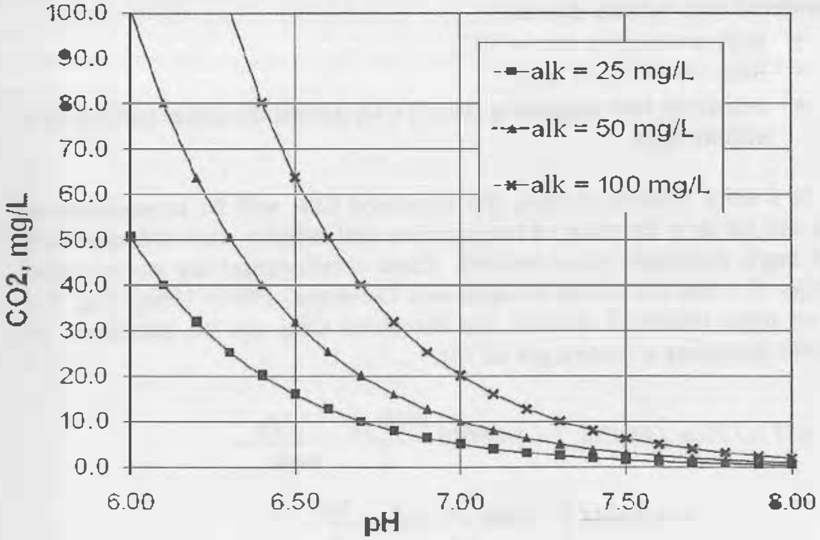


Figure 2.2 The relationship between pH, alkalinity, and CO_2 concentrations when alkalinity is 25, 50, & 100 mg/L at zero salinity and 20 °C (Note CO_2 concentration is proportional to alkalinity).

In recent years, as aquaculture system stocking density and hydraulic retention time has increased, the relationship between pH and alkalinity has become a significant issue. This relationship requires careful monitoring and adjustment of both alkalinity and carbon dioxide levels to maintain optimum pH for both the aquatic species being grown and the biofilters. Alkalinity is easily adjusted through the addition of sodium bicarbonate ($NaHCO_3$), common baking soda. Other materials can be used, but sodium bicarbonate is commercially available in 23 to 45 kg (50 to 100 lb) bags, safe, inexpensive, and easy to apply. It has very high

water solubility and rapidly dissolves in water at ambient temperature. A general rule of thumb is that for every pound of feed, approximately 0.25 lbs (113 g) of sodium bicarbonate should be added to the water. Carbon dioxide concentrations are routinely controlled through degasification systems, such as counter-flow gas stripping towers.

Hardness is the term used to describe the ability of water to precipitate soap, the harder the water, the greater the amount of soap that must be added to a given volume of water to get the same cleaning action. In chemical terms, hardness is defined as the total concentration of primarily calcium (Ca^{2+}) and magnesium (Mg^{2+}), iron, and manganese in terms of mg/L equivalent calcium carbonate ($CaCO_3$). In practical terms, hardness is measured by chemical titration. The total hardness of natural waters ranges from less than 5 to over 10,000 mg/L $CaCO_3$. Waters have traditionally been classified as soft (0–75 mg/L), moderately hard (75–150 mg/L), hard (150–300 mg/L), or very hard (> 300 mg/L).

Hardness is often confused with alkalinity, probability because both are commonly defined in terms of mg/L $CaCO_3$. In fact, if the alkalinity of the water originates from limestone, the concentrations of hardness and alkalinity can be similar if not identical. Conversely, many coastal-plain area groundwaters have high alkalinity and very low hardness concentrations. Aquifers in regions of basalt and granite often have waters of low total hardness and low alkalinity, because of the relative low solubility of these minerals. If this low hardness and low alkalinity water is used for aquaculture, the water must be "hardened" by adding dissolved calcium to support newly fertilized freshwater fish eggs and for calcification of larval skeletal structures. Calcium and magnesium also decrease the toxicity of dissolved metals. Recommendations for total hardness range **from 20 to 300 mg/L.**

Carbon Dioxide and the Carbonate Cycle

Carbon dioxide is highly soluble in water, but concentrations in pure water are low (0.54 mg/L at 20°C), due to its low concentration in the atmosphere (about 0.035% by volume). Most of the carbon dioxide in an aquaculture water column is produced by animal respiration and the decomposition of organic matter. The concentration of carbon dioxide in groundwater can range from 0 to 100 mg/L, depending on the biological activity **in the aquifer recharge zone.** The concentration of dissolved carbon dioxide in surface waters depends on the rate of respiration, photosynthesis, and gas exchange with the atmosphere.

Exposure to high carbon dioxide concentrations reduces respiration efficiency and decreases the tolerance to low dissolved oxygen concentrations. High levels of carbon dioxide in the water reduce carbon

dioxide excretion at fish gills. This, in turn, causes the CO_2 concentration in the fish blood to increase, lowering the blood plasma pH, which creates a condition called respiratory acidosis. When a fish is in this condition, the amount of oxygen that the blood hemoglobin can carry is reduced and respiration distress can occur, even with high concentrations of dissolved oxygen in the water. This is referred to as the Bohr-Root effect. An upper limit of 15–20 mg/L carbon dioxide is recommended as steady state maximum for finfish (see Table 2.2), although this recommendation is poorly supported by research. Colt (2006) states that water quality criteria for carbon dioxide cannot be made at this time, especially for warmwater species. As a management technique, higher concentrations (60–80 mg/L) have a narcotic effect on aquatic animals and are sometimes used on a temporary basis as an anesthetic to reduce handling stress and during treatment procedures. There has also been work to use high levels of carbon dioxide to force fish to swim through connecting pipes from one tank to another (The Freshwater Institute, personal communication).

DEFINITIONS

- CO_2 = carbon dioxide
- H_2CO_3 = carbonic acid
- HCO_3^- = bicarbonate ions
- CO_3^{2-} = carbonate ions

Carbon dioxide differs from oxygen, nitrogen, and other gases, because its concentration in water is determined both by a gas-liquid equilibrium relationship and by a series of acid-base reactions. Gas-liquid equilibrium influences the transfer of carbon dioxide between air and water and the acid-base reactions determine the chemical form in which dissolved inorganic carbon is present in water. Dissolved carbon dioxide concentration is therefore a function of pH and the total dissolved inorganic carbon ($CtCO_3$) present in water, i.e., carbon dioxide (CO_2), carbonic acid (H_2CO_3), bicarbonate ions (HCO_3^-), and carbonate ions (CO_3^{2-}). It is more correct to write $H_2CO_3^*$ for carbonic acid where $H_2CO_3^*$ is a composite carbonic acid concentration that includes true carbonic acid plus dissolved carbon dioxide or:

$$[H_2CO_3^*] = [H_2CO_3] + [CO_{2_{aq}}] \tag{2.5}$$

This composite carbonic acid concentration is in fact what is referred to as dissolved carbon dioxide. The acid-base equilibrium relationships between pH and carbon dioxide in freshwater and seawater aquaculture systems have been extensively reviewed and a summary of the carbonate acid-base system is presented in Table 2.7 (Grace & Piedrahita, 1994, Stumm and Morgan, 1981).

The carbonate system can be considered to be a volatile system or a non-volatile system depending on whether or not aqueous carbon dioxide is allowed to exchange and equilibrate with atmospheric carbon dioxide, volatile being one that has equilibrated with the atmosphere. RAS's with medium or higher fish densities (> 40 kg fish biomass per m^3) are considered non-volatile due to:

- high production rate of CO_2
- high solubility of CO_2
- relatively low degassing rate of CO_2 across the water surface in a culture tank.

In a fully volatile system, the dissolved CO_2 will be at equilibrium with the air as a function of temperature and salinity and will approach ~0.5 mg/L (example given below). These relationships are summarized in Figs. 2.3 and 2.4 (from Bisogni and Timmons, 1994). Using Fig. 2.4 for an open (volatile) system, the dissolved CO_2 can be calculated as follows assuming a system pH of 7.0:

$$pH = 7.0 \Rightarrow [H_2CO_3] = 0.00001 \frac{mole_{CO_2}}{L} \cdot \frac{44 g}{mole_{CO_2}}$$

$$= 0.00044 \frac{g}{L} \cdot 1000 \frac{mg}{g} = 0.44 \frac{mg}{L}$$

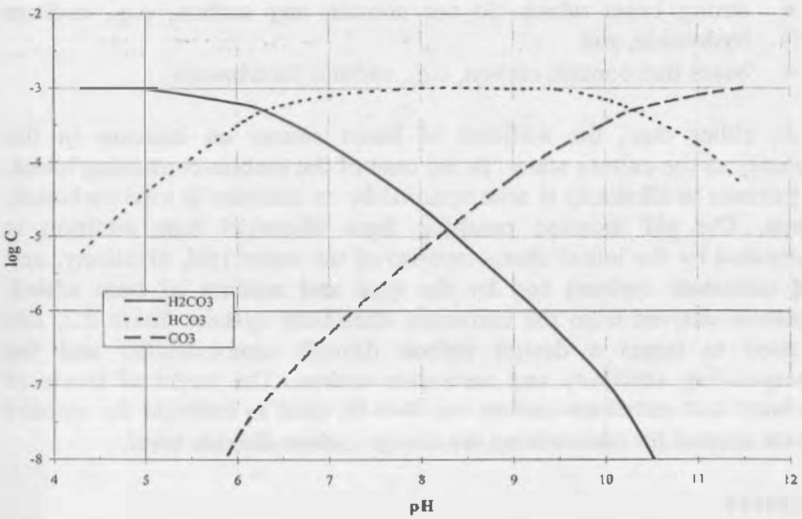


Figure 2.3 Non-volatile (closed) log C-pH diagram for freshwater at 20 °C (from Bisogni and Timmons, 1994).

Determination of equilibrium carbon dioxide concentrations requires that two of following three quantities be known: pH, total carbonate carbon, alkalinity. For example, total dissolved carbon dioxide, $[H_2CO_3^*]$, can be calculated from the total carbonate carbon, $[CtCO_3]$, and pH, since pH defines $[H^+]$:

$$\left[H_2CO_3^*\right]=\left[CtCO_3\right]\cdot\frac{1}{\left(1+\frac{K_0K_1}{\left[H^+\right]}+\frac{K_0K_1K_2}{\left[H^+\right]^2}\right)} \tag{2.6}$$

as well as from the alkalinity (ALK) and pH:

$$\left[H_2CO_3^*\right]=\left(\frac{ALK}{50000}-\frac{K_w}{\left[H^+\right]}-\left[H^+\right]\right)\cdot\left(\frac{1}{\frac{K_0K_1}{\left[H^+\right]}+\frac{K_0K_1K_2}{\left[H^+\right]^2}}\right) \tag{2.7}$$

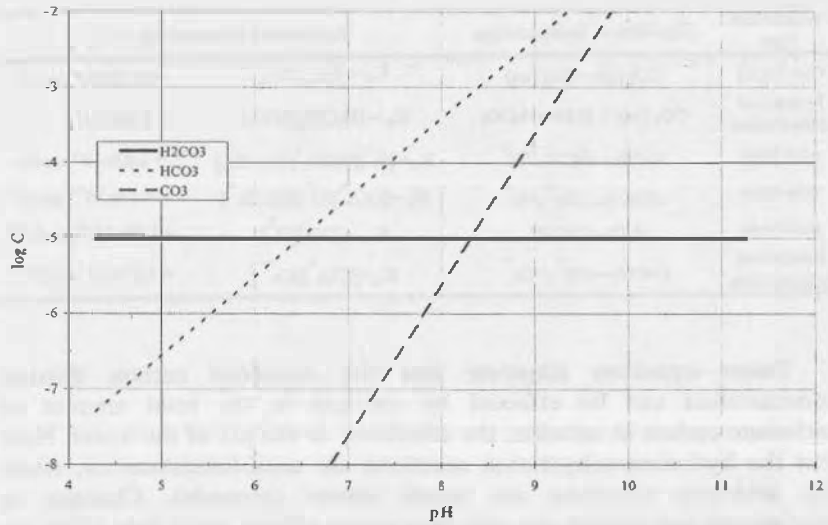


Figure 2.4 Volatile (open) Log C-pH Diagram for freshwater at 20 °C and for an atmospheric CO_2 concentration of 315 ppm (concentration of $10^{-3.5}$atm) (from Bisogni and Timmons, 1994)

The equilibrium constants K_0, K_1, and K_2 are defined in Table 2.7, the square brackets indicate molar concentration, and alkalinity is given as mg $CaCO_3$ per liter.

Applying the equilibrium constants given in Table 2.7 for CO_2 and assuming a gas space concentration of CO_2 around the stripping device of 5,000 ppm (OSHA 8-hr limit, see Table A-15), we can calculate the concentration of CO_2 in the water as follows:

$$X_{CO_2}=K_H\cdot P_{CO_2}=\frac{0.0006111\ mol\ CO_2}{atm\cdot mol\ water}\cdot 0.0050\ atm\cdot\frac{55.6\ mol\ water}{L}\cdot\frac{44g}{mol\ CO_2}\cdot\frac{10^3\,mg}{g}=7.5\frac{mg}{L}CO_2$$

Table 2.7 Carbonate System Acid-Base Equilibrium Reactions and Equilibrium Constants in Freshwater (Stumm and Morgan, 1981) (square brackets signify molar concentrations (moles/L); the units of the partial pressure of carbon dioxide (p_{CO_2}) are atmospheres (atm) (from Summerfelt et al. 2000; refer to Piedrahita and Seland, 1995, for temperature dependence of the equilibrium constants.)

Equilibrium Type	Equilibrium Relationships	Equilibrium Constants @ 25°C	
Gas-liquid	$CO_2(g) \leftrightarrow CO_2(aq)$	$K_H = P_{CO_2} / X_{CO_2}$	$= 6.11 \times 10^{-4}\ atm^{-1}$
hydration/ dehydration	$CO_2(aq) + H_2O \leftrightarrow H_2CO_3$	$K_0 = [H_2CO_3]/[CO_2]$	$= 1.58 \times 10^{-3}$
acid-base	$H_2CO_3 \leftrightarrow HCO_3^- + H^+$	$K_1 = [H^+][HCO_3^-]/[H_2CO_3]$	$= 2.83 \times 10^{-4}$ mol/L
acid-base	$HCO_3^- \leftrightarrow CO_3^{=} + H^+$	$K_2 = [CO_3^{=}][H^+]/[HCO_3^-]$	$= 4.68 \times 10^{-11}$ mol/L
acid-base	$H_2O \leftrightarrow OH^- + H^+$	$K_w = [OH^-][H^+]$	$= 1.00 \times 10^{-14}\ mol^2/L^2$
dissolution/ precipitation	$CaCO_3 \leftrightarrow CO_3^{=} + Ca^{++}$	$K_{sp} = [CO_3^{=}][Ca^{++}]$	$= 4.57 \times 10^{-9}\ mol^2/L^2$

These equations illustrate that the dissolved carbon dioxide concentration can be effected by changes in the total amount of carbonate carbon in solution, the alkalinity, or the pH of the water. Note that the hydration/dehydration reactions are semi-instantaneous, while the acid-base reactions are much slower (seconds). Changes in temperature and salinity can also have minor effects, since they affect the equilibrium constant values (Piedrahita and Seland, 1995). In addition to these equations, an approximation within 5–10% can be used to calculate the dissolved carbon dioxide concentration given the alkalinity and pH (restricted to a pH range from 6.5–9.5; Summerfelt, 1996):

$$C_{CO_2} = ALK \cdot 10^{(6.3 - pH)} \tag{2.8}$$

where:

C_{CO_2}	Dissolved carbon dioxide concentration (mg/L)
ALK	Alkalinity (as $CaCO_3$ mg/L)

Methods of CO_2 degassing are presented and discussed in Chapter 8. The addition of bases to increase the pH and shift the carbonate chemistry equilibrium will control carbon dioxide in aquaculture systems (Grace and Piedrahita, 1994; Summerfelt, 1996; Loyless and Malone, 1997). Typically adding bases to the culture water does not result in the removal of dissolved inorganic carbon from solution, but merely causes a decrease in the carbon dioxide concentration by shifting the carbonate carbon balance to bicarbonate and carbonate ions as the pH is increased.

Two general classes of chemicals are used in aquaculture to control pH and decrease carbon dioxide concentrations:

- strong bases which do not contain any carbon, e.g., sodium hydroxide, and
- bases that contain carbon, e.g., sodium bicarbonate.

In either case, the addition of bases causes an increase in the alkalinity of the culture water. In the case of the carbon-containing bases, the increase in alkalinity is accompanied by an increase in total carbonate carbon. The pH increase resulting from chemical base addition is determined by the initial characteristics of the water (pH, alkalinity, and total carbonate carbon) and by the type and amount of base added. Equations derived from the carbonate chemistry system, Table 2.7, can be used to target a design carbon dioxide concentration and the corresponding alkalinity and carbonate carbon. The target of levels of alkalinity and carbonate carbon can then be used to estimate the amount of base needed for maintaining the design carbon dioxide level.

SALINITY

Water is commonly described as fresh, brackish, or saltwater. Each of these terms refers to the salinity of the water, and there are no clearly identified crossover points between these characterizations. Salinity is defined as the total concentration of dissolved ions in water, and is usually reported as parts per thousand (ppt) or grams of salt per kilogram of water. The major contributors of dissolved ions are calcium, sodium, potassium, bicarbonate, chloride, and sulfate. The salinity of natural waters tends to reflect the climate, geography, and hydrological conditions of the immediate surroundings. Each aquatic species has an optimum range of salinity for reproduction and growth, although the salinity tolerance of most aquaculture species is rather broad. For example, rainbow trout fingerlings are produced in freshwater, acclimated to saltwater and then can be grown out to marketable size in sea cages at salinities as high as 20 ppt. Most freshwater fish of commercial importance reproduce and grow well at salinities up to at least 4–5 ppt.

Fish maintain the concentration of dissolved salts in their body fluids by regulating the uptake of ions from the environment and through the restriction of ion loss. This process is called "osmoregulation". Freshwater fish, for example, tend to accumulate water because they have body fluids more concentrated in ions than the surrounding water. When exposed to salinity values outside of their optimum range, aquatic species must spend considerable energy for osmoregulation at the expense of other functions, such as growth. If salinity deviates too far from optimum, the animal cannot maintain homeostasis and dies. The

blood of freshwater fishes has an osmotic pressure approximately equal to the osmotic pressure of a 7 ppt sodium chloride solution. Freshwater aquaculture systems are generally maintained at 2–3 ppt salinity to reduce stress levels and the amount of energy required for osmoregulation, thereby increasing growth rates.

SOLIDS – SETTLEABLE, SUSPENDED, DISSOLVED

Waste solids accumulating in an aquaculture system come from uneaten feed, feed fines, fish fecal matter, algae, and biofilm cell mass sloughed from biological filters. Studies indicate that fish produce between 0.3 to 0.4 kg total suspended solids (TSS) for every 1 kg of feed fed. Waste solids influence the efficiency of all other unit processes in a recirculating system. They are a major source of carbonaceous oxygen demand and nutrient input into the water, and can directly affect fish health within recirculating systems by damaging fish gills and harboring pathogens. The tentative upper limit for freshwater fish is 25 mg TSS/L, with 10 mg TSS/L usually recommended for normal operation; tilapia can perform well with TSS levels of 80 mg/L if all other water quality parameters are at a preferred level. Therefore, solids removal is one of the most important processes in aquaculture systems. Optimally, solids need to be removed from the fish culture tank as soon as possible, while creating as little turbulence and mechanical shearing as possible.

Solids are generally classified into three categories: settleable, suspended, and fine or dissolved solids. The difference between settleable and suspended solids is simply the time it takes for them to settle to the bottom of an Imhoff cone. Settleable solids settle out in less than an hour. Suspended solids do not; and therefore require a treatment process other than conventional gravity settling basins. Fine and dissolved solids are by their very nature difficult to remove.

2.6 MEASUREMENTS

METERS & INSTRUMENTS

The three water quality parameters described below are the three most critical to successful aquaculture, in that they can change dramatically in a relatively short time (DO can drop to lethal levels in minutes at high loading densities!). As a result, they should be monitored continuously, with backup systems and alarms to indicate out of tolerance conditions.

DISSOLVED OXYGEN

Although dissolved oxygen (DO) can be measured analytically by using the Winkler Method and a simple titration, the dissolved oxygen meters available today allow for rapid and accurate analysis over a range from zero to supersaturation. Two types of electrodes are commonly used: polarographic and galvanic. In simple terms, both of these meters consists of an electrode, which produces a signal proportional to the concentration of the oxygen in the water, and instrumentation to convert the signal reading to a visual display or recording device. A typical polarographic DO electrode or probe consists of a gold electrode and a silver-silver oxide reference electrode. The electrodes are bathed in 4 M KCl and separated from the sample by a membrane, usually made of Teflon, polyethylene, or fluorocarbon. The membrane is permeable to gases and the rate at which oxygen crosses the membrane is directly proportional to the dissolved oxygen concentration in the sample. When an electrical voltage is applied to the probe, molecular oxygen diffusing across the membrane reacts with the cathode (gold ring) and a small current flow to the anode electrode (silver). In a galvanic system, the electrode generates a small voltage (mV range) proportional to the dissolved oxygen concentration. Both systems require temperature, atmospheric pressure, and salinity compensation, usually accomplished by a combination of calibration and instrumentation hardware and software. Currently there are numerous DO meters available, over a wide range of price scales. As usual, you usually will "get what you pay for", so invest in a good oxygen meter (~ $500 minimum).

The newest technology for measuring dissolved oxygen is based upon dynamic fluorescence quenching of a luminescent dye (Luminescent Dissolved Oxygen, LDO). The sensor in the cap is coated with a luminescent material. Blue light from an LED illuminates the luminescent chemical on the surface of the sensor cap. The luminescent chemical instantly becomes excited and then as the excited chemical relaxes, it releases red light. The red light is detected by a photodiode and the time it takes for the chemical to return to a relaxed state is measured. The higher the oxygen concentration, the less red light is given off to the sensor and the shorter time it takes for the luminescent material to return to a relaxed state. The oxygen concentration is inversely proportional to the time it takes for the luminescent material to

return to a relaxed state (www.Hach.com). A simulated in-house EPA Tier 3 Validation Study (Jackson and Fair, 2004) results indicate the LDO sensor procedure is superior in performance to EPA Winkler reference method (EPA Method 360.2) and its alternative EPA approved membrane electrode method (EPA Method 360.1).

Unlike electrochemical dissolved oxygen sensor technologies, the LDO sensor does not consume oxygen. It does not require frequent recalibration or frequent cleaning (except when associated with consumptive slimes), resulting in longer sensor life and more stable and accurate readings. The system is also flow-independent so measurements can be made in applications with low or no flow.

Temperature

Traditionally, temperature was measured with a simple mercury thermometer. Recently, these are becoming harder and harder to obtain, due to the significant environmental harm (and potential economic hardship) it will do if broken and mercury is released into the environment (or in some cases, the production tank!). Thankfully, there are a host of alternative measurement devices, and most of them are more convenient and flexible in their use. To start with, most DO meters and pH meters have some form of built-in temperature measurement for calibration and temperature compensation. Second, simple handheld temperature meters are relatively cheap, accurate, and easy to use. Finally, these types of temperature measurements allow for easy data logging and control.

pH

Like the dissolved oxygen meters, easy to read pH meters are available over a wide range of specifications and price. In addition to pH, most pH

meters are also capable of measuring other ion specific electrodes, which include such parameters as ammonia, nitrate, Oxygen Reduction Potential, dissolved oxygen, conductivity, among others. However, please keep in mind that most of these ion specific electrodes were designed to be used under laboratory conditions and require some sample pretreatment, and they are limited to specific ranges of concentration. It is recommended that a combination, gel-filled pH probe be used. This instrument is inexpensive,

less susceptible to clogging, needs no refilling with electrolytes, and requires little or no maintenance.

The following parameters are usually slower to change than the parameters addressed above, and therefore can be monitored on a periodic (usually daily or weekly) basis. Of course, monitoring frequency can be increased if necessary due to a significant operational problem.

ORP (Oxygen reduction Potential)

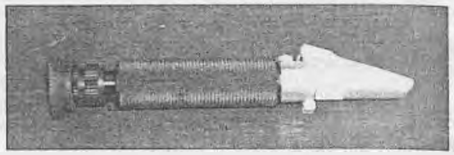
Oxidation Reduction Potential (ORP or Redox Potential) measures an aqueous system's capacity to either release or accept electrons from chemical reactions (Walker, 2009). ORP is used just like pH to characterize a systems water quality, but usually employed in aquaculture to control dosing of ozone. The ORP sensor works similarly to a pH probe using a two-electrode system that makes a potentiometric measurement expressed as either a positive or negative millivolts (mV). A solution with a positive ORP measurement will gain electrons (i.e. be reduced by oxidation) and a solution with a negative ORP will lose electrons (i.e. be oxidized by reducing the new species). Although measurement of ORP is relatively straightforward, differences between laboratory and in-plant sampling can vary significantly due to the effects of solution temperature and pH, the slow electrode kinetics, electrode poisoning, and the small exchange currents leading to drift and poor response. Still ORP measurements have proven useful as an analytical tool in monitoring changes in a system rather than determining their absolute value for example controlling the dosing of ozone.

Carbon Dixoide

Several types of dissolved carbon dioxide meters have recently become available. These work by first measuring the pH of the solution, and then calculating the CO_2 concentration based on the alkalinity (which must be known) and the carbonate equilibrium relationships. If the alkalinity is relatively constant, then CO_2 levels can be easily monitored and controlled by simply measuring the pH.

Carbon dioxide is usually measured by titration of a water sample to the phenolphthalene endpoint (pH 8.3) with a standard base. Approximate CO_2 concentrations can be determined from known values of pH, temperature, and alkalinity using either a formula or table values (see Fig. 2.2).

SALINITY



Salinity can be indirectly measured using the measurement of a physical property such as refractive index, conductivity, or density. Of these, the two most commonly used in aquaculture are the refractive index and conductivity measurements. A refractometer utilizes a change in the refractive index as function of the salt concentration to directly display the salinity level. It is very simple to use. Simply place a drop of water on the window and look through the eyepiece to directly read the salinity in parts per thousand. Refractometers provide fast, accurate salinity measurements, but some form of temperature-compensation is recommended when used outside their normal operating range of 20°C. One problem they do have is that most commercially available models have a wide salinity range of 0–100 ppt, making accurate measurements at low salinities difficult.

Electrical conductivity measurements can also provide a simple and quick measurement of salinity. Conductivity meters measure the flow of electrical current between two electrodes submersed in water, which is directly proportional to the concentration of ions, i.e., salts, in the water. They can be very accurate over a wide range of concentrations; these meters are direct reading and very portable.

Hydrometers measure salinity based on changes in water density (specific gravity) as a function of salinity. However, accurate and reliable hydrometers are more expensive than refractometers or electrical conductivity meters that provide the same degree of accuracy and ease of use, so they are typically not used. Inexpensive hydrometers are not recommended at all except for very gross estimates.

CHEMICAL ANALYSIS

Almost all of the following chemical analyses are easily performed colorimetically with off-the-shelf analysis kits manufactured by several companies. In most cases, using these kits is an easy process. Reagents come premixed in small vials, capsules, or bags, which requires only opening, mixing, and waiting for the specified period. Then, the mixture is measured, usually with a spectrophotometer. Analysis is quick, cheap, requires very little expensive laboratory equipment, and does not generate large quantities of hazardous wastes. Table 2.8 provides a summary of methods.

DISSOLVED OXYGEN (DO)

In the basic Winkler method, a sample of water is treated with manganous sulfate, potassium iodide, and sodium hydroxide. Under these highly alkaline conditions, the manganous ion is oxidized by the dissolved oxygen to manganous dioxide. Sulfuric acid is then added to dissolve the precipitate and produce acid conditions for the oxidation of iodide to iodine by manganous dioxide. The quantity of iodine released is proportional to the amount of dissolved oxygen (DO), and is estimated by titration with standard sodium thiosulfate with a starch indicator as the endpoint. This sounds difficult, but in practice, all of the reagents are available in a premixed form in many commercially available test kits and thus DO measurement is a routine activity. With the availability of relatively low cost dissolved oxygen meters, this method is usually used only for cross checking and calibration purposes.

<u>Testing Procedures for DO Transfer</u>. Standard tests for aerators are conducted using clean tap water at standard temperature and pressure (20°C and 760 mm Hg). Tests are based upon using deoxygenated water that has had the DO removed using a sodium sulfite (Na_2SO_3) solution using cobalt chloride ($CoCl_2{\cdot}6H_2O$) as a catalyst. The theoretical requirement for deoxygenation is 7.88 mg/L of sulfite (as Na_2SO_3) per 1.0 mg/L of O_2 removed. The cobalt chloride should be used at a concentration of 0.10–0.50 mg/L (as Co). A slightly higher concentration may be required if the water temperature is below 10°C. For more details, see APHA (1995).

CO_2

The simplest method of determining carbon dioxide concentration in the water column is to use a nomograph (see Standard Methods) when the pH and total alkalinity are known. Then, these values can be compared to the nomographic data to determine the CO_2 concentration of the sample.

If pH and alkalinity are not known, CO_2 can be determined by adding a titrating solution of sodium hydroxide (0.0227 Normality or N) to a

specified volume of sample water (normally, 250 mL). Since water at a pH over 8.34 does not contain appreciable amounts of carbon dioxide, the amount of a base (normally, sodium hydroxide) needed to raise the sample's pH to the 8.34 pH endpoint is approximately equivalent to the carbon dioxide content of the sample. Free carbon dioxide reacts with sodium to form sodium bicarbonate, which raises the sample's pH. Thus, sufficient amounts of the titrating solution of sodium hydroxide are added to raise the sample's pH to 8.34. The quantity of sodium hydroxide that was required to reach this endpoint is equal to the amount of CO_2 in the sample, because 1 mL of sodium hydroxide (0.0227 N strength titrating solution)) equals 1 mg/L CO_2 concentration in the sample.

ALKALINITY

Alkalinity is a measure of the acid neutralizing capacity of the water. Alkalinity can be measured by titration with 0.02 N sulfuric acid and titrating to a pH end-point or using a phenophalein indicator to show the color change as the indicator to stop adding titrant. With a 0.02 N sulfuric acid, one mL of titrant equals 10 mg/L of alkalinity (defined as calcium carbonate, $CaCO_3$). Only a few items are required to perform these measurements: a 125 mL beaker, a 50 mL buret, 0.02 N sulfuric acid (available premixed), methyl orange indicator, and a dropper.

Table 2.8 Water Quality Parameters: Standard Methods

Water Quality Parameter	Standard Methods Monitoring method
Temperature	2550 Temperature Temperatures are traditional made with mercury-filled thermometers, but non-mercury or electronic temperature meters are prefered due to the toxicity of mercury in the environment.
pH	pH Meters Follow manufacturer's instructions for calibration using known standards 4.0, 7.02 and 10.
Total Suspended Solids	2540 Solids A well-mixed sample is filtered through a weighed standard glass-fiber filter and the residue retained on the filter is dried to a constant weight at 103 to 105 °C. The increase in the weight of the filter represents the total suspended solids.
Ammonia	4500 – NH_3 Nitrogen (Ammonia) Colorimetric Nesslerization Method: nessler-ammonia reaction produces a yellow to brown color, measured with spectrophotometer at 425 nm.
Nitrite	4500-NO_2^- Nitrogen (Nitrite) Colorimetric method with diazotized sulfanilamide with NED dihydrochloride creating a reddish purple azo dye measure with a spectrophotometer at 543 nm.
Nitrate	4500-NO_3^- Nitrogen (Nitrate) Cadmium reduction to nitrite and measurement of NO_2^-.
Phosphorus	4500-P Phosphorus (Orthophosphate) Ammonium molybdate and potassium antimonyl tartrate react to form a heteropoly acid, which is reduced to an intensely colored molybdenum blue by ascorbic acid.
Alkalinity	2320 – Titration Method Titration with 0.02 N Sulfuric Acid with methyl orange indicator end point (4.5 pH), 1 mL titrant equals 10 mg/L $CaCO_3$.
Hardness	2340 Hardness EDTA titrimetric method using EDTA and ethylenediaminetetraacetic acid and Calmagite. Titrated with EDTA from a red to a blue color.
Carbon Dioxide	4500-CO_2 Carbon Dioxide Free CO_2 reacts with sodium hydroxide (0.0227 N) to form sodium bicarbonate; completion indicated using a pH meter (8.3) or phenolphthalein indicator. 1 mL of NaOH equals 1 mg/LCO_2.
Salinity	2520 Salinity Measurement of conductivity, density, or refractive index. Commercially available conductivity meters and refractometers.
Chlorine	4500-Cl Chlorine (Residual) Iodometric method, chlorine liberates iodine from potassium iodide solutions and the iodine is titrated with a standard solution of sodium thiosulfate with starch as the indicator.
Total Gas Pressure	2810 Dissolved Gas Supersaturation Commercially available membrane-diffusion instruments.
ORP – Oxidation-Reduction Potential	2580 Oxidation-Reduction Potential (ORP) ORP Electrode and standard reference electrode. Follow manufacturer's instructions for calibration using known standards.
Conductivity	2510 Conductivity Conductivity is a measure of the ability of an aqueous solution to carry and electric current. Standard meters available, calibrate with a standard KCL solution.

AMMONIA/NITRITE/NITRATE

The easiest method for measuring ammonia, nitrite, and nitrate is to use a colorimetric technique and either a color wheel comparison or a spectrophotometer.

For ammonia, the colorimetric Nesslerization method is the most popular. In this method, a nessler-ammonia reaction produces a yellow to brown color that is proportional to the ammonia-nitrogen concentration. This is a simple process to use, requiring only the addition of one commercially available reagent to the water sample, and then taking a measurement of the ammonia-nitrogen concentration with a spectrophotometer at 425 nm.

Nitrite is usually measured using a Colorimetric method. Diazotized sulfanilamide and NED dihydrochloride are added to the water sample, which creates a reddish purple azo dye, which is then measured with a spectrophotometer at 543 nm. The degree of transmittance in the spectrophotometer correlates to nitrite concentration data as presented in the form of standardized curves. Finally, nitrate is measured by reducing it to nitrite with a Cadmium catalyst and then measuring nitrite concentration.

Reagents for all of these analysis methods are readily available in premixed or packet form. There are also several manufacturers of spectrophotometers, including Hach, YSI, and LaMotte. Although the use of a color wheel comparison is applicable in some cases, for commercial systems the cost of a spectrophotometer and the flexibility and accuracy it provides is worth the expense.

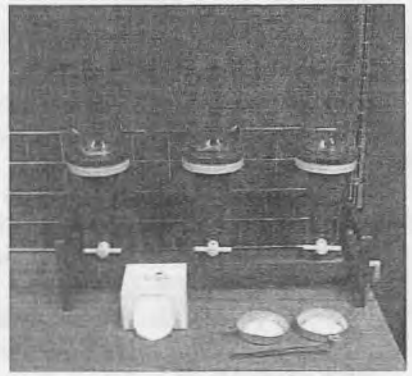
Ion Specific Electrodes (ISE) allows measurement of ammonia, nitrate, and several other parameters using the pH/mV input of a standard pH meter. Although these have been used in the wastewater treatment industry and in a laboratory setting, they are not popular in the

aquaculture industry. This is probably because of the difficulty in maintaining calibration, the need to pre-treat the water sample, and high equipment costs associated with this type of measurement method.

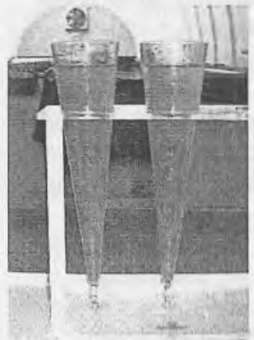
PHOSPHORUS

Phosphorus is measured by using a colorimetric technique similar to ammonia-nitrogen. In this method, ammonium molybdate and potassium antimonyl tartrate are added to the sample, and they react to form a heteropoly acid. Then, ascorbic acid is added, which reduces the heteropoly acid to intensely colored molybdenum blue. The reactive phosphorus concentration (orthophosphorus) is then measured with a spectrophotometer at 880 nm.

SOLIDS



Total solids (TS) are the weight of the material residue left after evaporation of a sample at a temperature of 105°C. Total suspended solids (TSS) are the portion of material retained by a filter, and total dissolved solids (TDS) are the portion of material that passes through the filter. Total volatile solids (TVS) is the difference in weight of the total solids after burning them to ashes, and Settleable solids (SS) is the material that settles out of suspension within a defined period of time.



The most commonly used measurement of solids is the total suspended solids and the settleable solids. The amount of total suspended solids is simple to determine, although a fair amount of laboratory equipment is required. The equipment requirements are: a drying oven (105°C), a filtration setup including vacuum pump, an analytical balance (0.1 mg), and glass fiber filters (Whatman GF/C). To take a TSS measurement, first weigh the glass fiber filter, and then filter a known volume of sample through it. Next, dry the filter at 105°C for a least one hour. When dry, weigh the filter again and divide the difference in

weight by the volume of sample. The result is the total suspended solids concentration (mg/L).

Settleable solids are measured with an Imhoff cone, which is a cone shaped container with volume gradations at its base. A sample of 1 L is poured into the Imhoff cone and allowed to settle for one hour, after which the volume of settleable solids in the base of the cone are read in mL/L.

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CHAPTER 3
MASS BALANCES, LOADING RATES, AND FISH GROWTH

3.0 INTRODUCTION

Water flow is the mechanism by which oxygen is transported into a fish culture vessel and the waste products being generated within are removed. The design of a recirculating aquaculture system (RAS) should insure that the important parameters affecting water quality and fish productivity, e.g., oxygen, ammonia, carbon dioxide, and suspended solids are properly balanced. This requires calculating the value of each of these parameters independently to determine the thresholds for each. Then, having done the necessary calculations, the system must be operated at the lowest flow rate possible while still maintaining a particular parameter at its design value, e.g., ammonia. Obviously, the minimum flow rate possible while maintaining one particular parameter may be too low for maintaining another. The same mass balance approach can be utilized on any variable affecting water quality. It simply comes down to balancing the transport in, the production of a particular parameter within the culture tank, and the transport out. In word equation form, engineers like to say:

$$\textit{Transport in of "x"} + \textbf{\textit{production}}\textit{ of "x"} = \textit{transport out of "x"} \quad (3.1)$$

The production term can be the production of oxygen, ammonia, suspended solids, or CO_2. Note that the production term can be negative, meaning consumption of a certain component, e.g., oxygen.

Figure 3.1 depicts a mass balance for the general case where part of the flow is recirculated and part of the flow is flow-through.

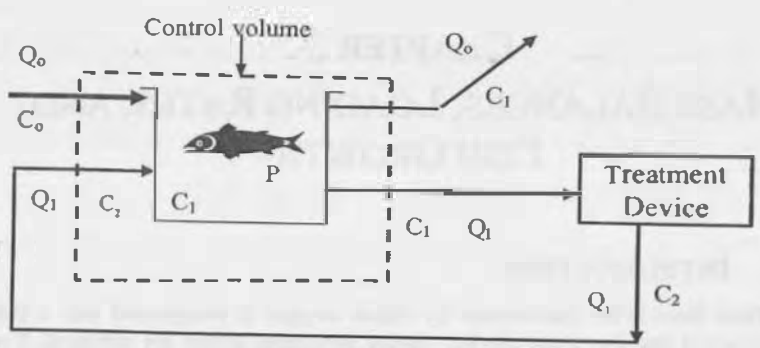


Figure 3.1 General mass balance on a fish culture tank. Treatment occurs exterior to the tank.

This assumes a completely well mixed tank and that the tank has reached a non-changing condition, i.e. equilibrium, with respect to time or steady-state conditions. The box outside the fish tank represents some treatment device or process that changes the concentration of the noted parameter "x". (Note: there could be several treatment devices, each treating a different water quality variable.)

Returning to our word equation, Eq. 3.1 and Fig. 3.1 the mass balance equation can be applied assuming steady state conditions:

$$Q_1 C_2 + Q_0 C_0 + P = Q_0 C_1 + C_1 Q_1 \qquad (3.2^a)$$

To obtain accurate and reliable results from these equations, it is essential that each of the terms or products of terms in Eq. 3.2 is represented by the same unit value, e.g.,

$$\frac{kg_{oxygen}}{day}$$

For example, the unit balance example for a transport of oxygen flowing into the tank would be:

$$QC = \frac{kg_{water}}{day} \frac{kg_{oxygen}}{kg_{water}} = \frac{kg_{oxygen}}{day} \qquad (3.3)$$

[a] Symbols are defined at end of chapter.

If we were doing a mass balance using Eq. 3.2 for oxygen, then all terms or products would need to have the same units of kg oxygen per unit time. It is convenient to use "day" as the time unit, since growth rates and feeding rates are generally measured on a per-day basis. BE CAREFUL to be consistent with unit designations.

Transport is the key term in these calculations, and it is defined as the product of flow and concentration. For example, the remainder of oxygen transport into the tank minus the allowable minimum level of oxygen departing the tank defines the oxygen available for fish growth. Flow is measured as volume per time or mass per time, and will be usually defined in terms of::

- gallons per minute, gpm
- liters per minute, Lpm
- kg per sec, kg/s
- cubic meters per sec, m^3/s

Typically, most water quality parameters are expressed in terms of::

$$\frac{milligram}{Liter} \quad or \quad \frac{mg}{L}$$

The usage of mg/L is often called or referred to as:

ppm or parts per million.

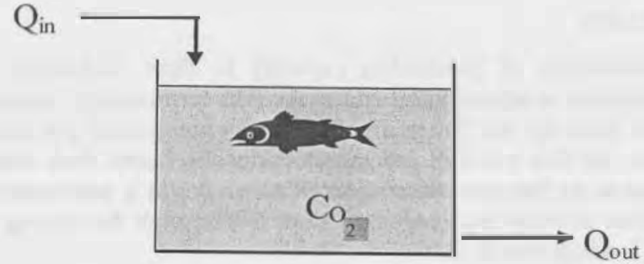
These values are the same.

This usage comes from:

$$\frac{mg}{L} \cdot \frac{L}{1000\,g} \cdot \frac{g}{1000\,mg} = \frac{1}{10^6} \quad or \quad \frac{1\ part}{million} \text{ or } 1\,ppm$$

Thus: $$10\frac{mg}{L}\ oxygen \rightleftarrows 10ppm\ oxygen$$

Now, to reinforce this concept, let's calculate the available oxygen to support fish growth assuming a mass flow of water of 100 gpm of saturated inlet water at 60°F at an elevation 800 feet above sea level.



$Q_{in}\,C_{in}$ = O_2 transported in
$Q_{out}\,C_{tank}$ = O_2 transported out

Equation 3.4 provides an approximation that can be used to estimate oxygen concentration in non-saline waters (Chapter 10 provides a more detailed approach to determine gas solubility including oxygen, carbon dioxide, nitrogen, and argon):

$$C_{saturation}\frac{(mg)}{L}=\frac{132}{(T^{\circ}F)^{0.625}}\cdot\frac{760}{760+\dfrac{altitude(ft)}{32.8}} \tag{3.4}$$

Using Eq. 3.4:

Altitude of 800 ft.
Temperature 60°F

$$C_{sat}=10.21\cdot 0.97=9.89\frac{mg}{L}$$

For the mentioned flow rate of 100 gpm of water and with the incoming water having a concentration of 9.89 mg/L, the mass of oxygen transported into the tank on a daily basis is ("gpm" units are used since they are common terminology in US):

$$Q_{in}\,C_{in,\,oxygen}=100\,\frac{gal}{min}\cdot 9.89\,\frac{mg}{L}$$

Now, make the units consistent:

$$=100\,\frac{gal}{min}\cdot 3.785\,\frac{L}{gal}\cdot 9.89\,\frac{mg}{L}\cdot 1440\,\frac{min}{day}=\frac{mg}{day}\,O_2$$

$$=5{,}390{,}000\,\frac{mg}{day}\cdot\frac{kg}{10^6\,mg}=5.39\,\frac{kg\ O_2}{day}$$

This then is the oxygen available to the culture vessel on a daily basis to support fish growth and bacterial demand. However, the water leaving the tank must still be at the minimum level necessary to support fish growth, e.g., 5 mg/L, so only a portion of the total available oxygen can be used.

The available oxygen is then (same as previous example except now a C_{out} for the discharge water is defined):

$$Q\,(C_{in}-C_{out})=$$

$$=100\,\frac{gal}{min}\cdot 3.785\,\frac{L}{gal}\cdot(9.89-5.0)\frac{mg}{L}\cdot\frac{kg}{10^6 mg}\cdot 1440\,\frac{min}{day}$$

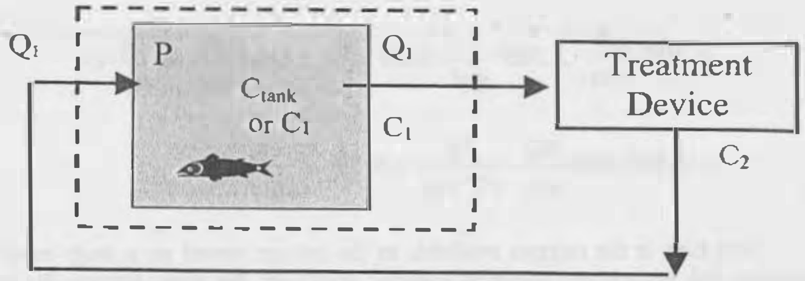
$$=2.66\,\frac{kg}{day}\,O_2$$

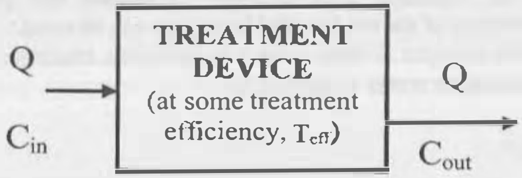
Returning to the general mass balance Eq. 3.2, the simplest RAS case is where all water flow is recirculated and there is no discharge. In this situation, the Q_0 terms in Eq. 3.2 drop out $Q_{in}=Q_{out}$ have:

$$Q_1\,C_2+P=+\,Q_1\,C_1 \tag{3.5}$$

$$Q_1\,(C_2-C_1)=-P$$

Solving the general mass balance equation with the "magical" treatment box, we need to determine the concentration of each particular water quality parameter leaving the treatment device (C_2), so that the mass flow of the water quality parameter into the fish culture tank can be determined and the mass balance solved. Looking now at only the treatment device depicted in Fig. 3.1, the treatment box could be a biofilter, a CO_2 stripper, or a solids settling chamber. Each will have its own treatment efficiency for the particular water quality parameter it is designed to treat. Engineers just use the box as a symbolic depiction of this treatment device.





Looking at this treatment box and doing a mass balance on the control volume, you can solve for the concentration of the water quality parameter of interest, C_{out}. Since the water flow in equals the water flow out, the C_{out} term can be solved for directly:

$$C_{out} = C_{in} + \frac{T}{100}\left(C_{best} - C_{in}\right) \tag{3.6}$$

where, C_{best} is the absolute best result obtainable by a treatment system, e.g., zero ammonia or saturated oxygen, zero suspended solids.

The term "C_{best}" has been adapted to represent what the treatment device is trying to do. If you had the perfect treatment device, the device is still limited by basic physical laws as to what it can achieve. Thus, the reference to *best*. Note that if the device is an oxygen addition unit, the C_{best} term can be increased above atmospheric concentration values for oxygen by increasing the partial pressure above atmospheric oxygen partial pressure in the device. For example, a pure oxygen device will have a C_{best} value of roughly five times the C_{best} value that is available if normal air used at atmospheric pressure, e.g., trickling tower. C_{best} for most other parameters should be fairly obvious to the reader, e.g., ammonia and TSS are zero, but CO_2 will be around 0.5 mg/L since there is some CO_2 in the air.

Loading Rates

The relationships of production capacity to flow exchanges and space are important to aquacultural engineers. The terminology "loading" (L) is used to describe the fish mass that can be maintained per unit of flowing water, kg fish per liter per minute flow (kg/Lpm). Fish density (D_{fish}), defined as kg fish per cubic meter of space (kg/m^3), and combined with the number of water exchanges per hour (R) through the rearing unit produces the loading rate (L):

$$L = 0.06\frac{D_{fish}}{R} \tag{3.7}$$

The constant 0.06 in Eq. 3.7 converts Lpm to m^3/hr (1.0 Lpm · 60 min/h = 60 L/h or 0.06 m^3/h, there are 1,000 L in a m^3). The loading capacity depends primarily on water quality, fish size, and species. Using Eq. 3.7 and assuming that fish metabolism requires 250 grams of oxygen for each kg of feed fed (no allowance for nitrification), the allowable loading (kg of fish per Lpm of flow) due to oxygen constraints is:

$$L_{oxygen} = \frac{144\,\Delta O_2}{250 F_{\%}} \tag{3.8}$$

The ΔO_2 term in Eq. 3.8 represents the allowable drop in oxygen from inlet to outlet. For example, if the inlet DO is 11.0 mg/L and the outlet is designed to maintain oxygen above 5.0 mg/L, then the ΔO_2 is 6.0 mg/L. Eq. 3.8 is demonstrated in Fig. 3.2 for various feeding rates and allowable changes in oxygen concentration.

As fish densities increase, minimum oxygen levels are better maintained by using pure oxygen systems, which permit inlet oxygen concentrations to be maintained at multiples of dissolved atmospheric concentration levels (see Chapter 10). However, other water quality parameters will begin to limit the allowable carrying capacity due to degradation in accumulated ammonia or carbon dioxide or suspended solids. The term for this condition is the cumulative oxygen consumption (COC) level. From basic stoichiometry and the production values for ammonia, solids, and carbon dioxide associated with feeding, then:

10 mg/L of oxygen consumed will produce up to:

- 1.4 mg/L of ammonia,
- 14 mg/L of carbon dioxide, and
- 10 to 20 mg/L of suspended solids.

These relationships must be kept in mind when designing the fish culture vessel.

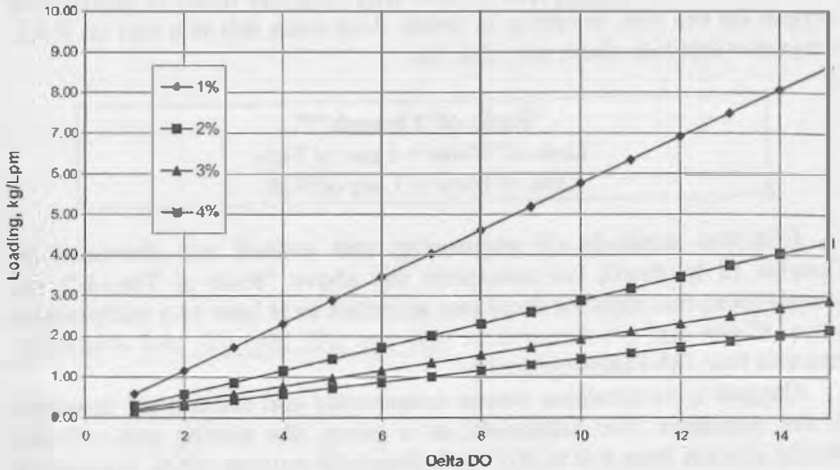


Figure 3.2 Allowable Fish Loading in kg per Lpm. Based upon the change in dissolved oxygen from inlet to outlet (Delta DO, mg/L) and the percent body weight per day for the fish feeding rate (to convert kg/Lpm to lb per gpm, multiply by 8.33).

Maximum allowable or safe densities (D_{fish}) are much more difficult to ascertain than are maximum loadings. It still seems to be a very subjective process and has much controversy. Density is primarily a function of fish size, species, and the characteristics of the rearing environment and management skill. New growers tend to overestimate their own safe loading densities and assume they can establish and sustain densities from the very beginning that in fact require expert management skills. Do NOT fall into this trap. You will kill fish. New growers should target about 1/2 the densities recommended in this book for expert growers. This subject is discussed in detail in Chapter 4, Culture Tank Design.

3.1. Production Terms

The "P" term in Eq. 3.2 represents the production of some pollutant or consumption term. These terms in an RAS system can all be proportionately related to the fish feeding rate. In principle, if you are not feeding the system, there is no pollution. This generalization is valid, because even ammonia production rates reduce 10 fold within a day or so, once feeding activity has ceased. Oxygen consumption is also reduced by approximately 50% and fecal production goes to zero. However, the idea in any production system is to grow the animals, and to grow, they must be fed, so we relate the production terms exclusively to feeding rates as follows::

P_{Oxygen} = - 0.25 kg per kg feed consumed by fish (a negative production term)

- 0.12 kg per kg feed consumed by nitrifying bacteria

- 0.13 heterotrophic bacteria (estimate, can be as high as 0.5)

P_{Oxygen} = - 0.50 kg per kg feed for system

(including nitrifying and heterotrophic bacteria, this is a MINIMUM VALUE; safe design value is 1.0 kg oxygen per kg feed fed)
(Note: see alternative method of estimating oxygen in Section 3.2 Oxygen)

P_{CO2} = 1.375 grams produced for each gram O_2 consumed

(both fish and bacteria))

P_{TAN} = F · PC · 0.092

P_{TSS} = 0.25 · kg feed fed (dry matter basis)

(literature gives values from 20% to 40% of feed fed on dry basis)

3.2 WATER QUALITY DESIGN TARGETS

A difficult concept to grasp for most is that in performing the mass balances, the designer/manager must choose design or target operating conditions. These are the "C" values in Eq.3.2 shown in the culture tank. These design numbers are species dependent and are continually being refined for RAS applications. The authors' recommendations are given in Table 3.1 for a common warm (tilapia) and cool (trout) water species.

Table 3.1 Water Quality Parameters for a Cool and Warm Water Species

Parameter	Tilapia	Trout
Temperature, °C (°F)	24 to 30 (75 to 85)	10 to 18 (50 to 65)
Oxygen, mg/L	4 to 6	6 to 8
Oxygen partial pressure, mm Hg	90	90
CO_2, mg/L	30 to 50	20 to 30
Total suspended solids, mg/L	<20	<10
Total ammonia - N, mg/L	<3	<1
NH_3-N, mg/L	<0.06	<0.02
Nitrite-N, mg/L	<1	<0.1
Chloride, mg/L	>200	>200

SELECTING TARGET VALUES FOR WATER QUALITY

Calculating the minimum flows required to maintain targeted values for water quality (and then using the largest minimum value found for all the different water quality variables) will show how sensitive the calculated flow rates are to the value selected for the design value. A typical scenario is to select a value, do the calculations, realize that there is no way you could afford to supply such a high flow rate, and then start to make adjustments in the targeted values, e.g., 4 mg/L oxygen is probably OK instead of the 6 mg/L you originally chose, etc, etc. In the end, one must choose realistic values and then stay with these choices and the ramifications of the resulting flows required to maintain the mass balances. Do NOT ever compromise on the required flow rates. You will be sorry if you do.

OXYGEN

The major reason most fish die is from lack of oxygen due to a loss of water flow. This is because oxygen is consumed at a fairly high rate (fish metabolism) and oxygen is transported by water flow. Due to low inherent concentrations of oxygen, "high" flows are required to transport the required oxygen. Flows required to maintain a satisfactory oxygen level are generally the controlling flow rate parameter when solving the

series of mass balance equations to determine the most restrictive parameter. Even a partial loss of flow will generally result in insufficient oxygen for the fish, resulting in death. And since this is a text on RAS, remember this rule about why fish die:

"Rule of Thumb"
Loss of Water = Loss of Fish
Loss of Flow = Loss of Fish

Effective methods of monitoring and control are discussed in Chapter 13 in detail, but remember the above "Rule of Thumb"; so, always sense and monitor these two activities in at least two independent ways. If you don't, we guarantee that you will lose fish and eventually you will lose fish catastrophically.

Oxygen concentrations versus temperature and salinity are provided in the Appendix. For salmonids, as a group, the rearing unit effluent should contain from 6.0 to 8.0 mg/L dissolved oxygen (DO). For catfish and tilapia, allowable minimum levels are much lower than for salmonids, e.g., 2 or 3 mg/L, while it is certainly recommended to stay much closer to 5 or 6 mg/L. This variation in what the allowable minimums are has to do with the fact that partial oxygen pressure (pO_2) appears to be a more valid way to determine the lower limits. A pO_2 of 90 mm Hg seems to be a reasonable target for salmonids (Downey and Klontz, 1981). The atmosphere contains 21% oxygen, and at standard pressure of 760 mm Hg at 20°C, this represents a pO_2 of 0.21 • (760–17.54)[b] or 155.9 mm Hg. and corresponds to a dissolved oxygen saturation of 9.1 mg/L. Therefore, 90 mm Hg corresponds to 5.2 mg/L DO. If the temperature is 5°C, pO_2 corresponds to 0.21 • (760 – 6.54) or 158.2 mm Hg and corresponds to a DO saturation 12.8 mg/L. In this case, 90 mm Hg corresponds to 7.3 mg/L. As noticed, the warmer the water the lower the effluent DO in mg/L can go, while still representing a pO_2 of 90 mm Hg. Applying 90 mm pO_2 @ 30°C for tilapia would set a target value of 4.4 mg/L for culture tank oxygen levels (consistent with Table 3.1).

For salmonids and using 90 mm Hg as the minimum target, the available oxygen at 5°C is 5.8 mg/L (12.8–7.0 mg/L), while at 20°C it is only 2.1 mg/L (9.1–7.0 mg/L), or more than a 60% reduction. Thus, considerably less oxygen is available at higher temperatures when metabolic rates, too, are higher, significantly impacting production capacity on the basis of limited available oxygen. Note the dilemma between higher temperatures supporting higher fish

[b] 17.54 mm Hg is the vapor pressure of water at 20°C

growth and metabolism, but the increasing unavailability of oxygen in the water column to support growth.

Beyond the general rule that each unit of feed will require 0.25 units of oxygen for fish metabolism, usage rates will depend mostly upon the type of fish being considered. Westers (1979) uses 200 to 250 g per kg of feed fed, while Pecor (1978) recommends using 110 g per kg of feed fed for esocids, such as the tiger muskellunge, a coolwater, non-active fish. These values are consistent with our general recommendation of 250g O_2 per kg of feed fed for fish oxygen needs. Bob Robinson (Fish Farming News Issue 6, 2009) gave the following guidelines for estimating oxygen use by fish.

Warm water: Juveniles and Adults	0.05	0.03 kg/hr/100 kg fish
Cool water: Juveniles and Adults	0.04	0.025
Cold water: Juveniles and Adults	0.03	0.02

The above estimates are easy to apply and can then be compared against the P terms given in Section 3.1 that are based upon feeding loads and bacteria oxygen usage and then you should use the higher of the two values to estimate your design requirements.

Finally, remember that not only do the fish require oxygen, but also the biological filter is just as critically dependent upon adequate oxygen levels to support bacteria metabolism. The DO concentration within the filter must be maintained at or above 2.0 mg/L to insure that the rate of nitrification in the filter does not become limited because of oxygen depletion (Kumar, 1984; Manthe et al. 1988). Always measure the DO coming off the biofilters and if the concentration starts to approach 2.0, then take corrective action, e.g., increase flow rate through the biofilter by increasing the hydraulic loading rate on the filter.

AMMONIA

There is considerable confusion about ammonia. Definitive values for the toxic levels of ammonia and the differentiation between the toxic NH_3 form and the supposed non-toxic NH_4^+ have never been determined. Meade (1985) reviewed the published literature on the effects of ammonia on fish and concluded:

> *A truly safe, maximum acceptable concentration of un-ionized, or of total ammonia, for fish culture systems is not known.*

The apparent toxicity of ammonia is extremely variable and depends on more than the mean or maximum concentration of ammonia.

The European Inland Fishery Advisory Commission (EIFAC) of FAO has set 0.025 mg/L as the maximum allowable un-ionized ammonia nitrogen (NH_3–N or $A_{NH3\text{-}N}$). Note, this means that tank ammonia levels (TAN) can exceed 10 mg/L if pH is maintained below 7.0. We would be very uncomfortable using such a high design value. What if the pH rose to 7.3 and doubled your NH_3? As seen in Table 3.1, we use rule of thumb values of 1 mg/L TAN for cool water and 2 or 3 for warm water fish. You should always check your TAN target value selection by assuming some pH that you intend to maintain and see if your NH_3 concentrations will exceed the 0.025 mg/L value using the ammonia-ammonium pH Temperature tables from the Appendix. If so, rethink your design carefully before proceeding.

There seems to always be a disproportionate amount of attention applied to nitrification and ammonification. All these calculations depend upon the estimate of an ammonia generation load, which is based upon the fish feeding rate:

$$P_{TAN} = \frac{F \cdot PC \cdot 0.092}{t} \quad (3.9)$$

where 't' is usually one day, i.e. feed fed uniformly over a 24 h period.

The constant in the ammonia equation is based upon a series of approximations and estimates that when multiplied together result in the 0.092.

0.092 = 0.16 * 0.80 * 0.80 * 0.90

- 16% (protein is 16% nitrogen)
- 80% nitrogen is assimilated
- 80% assimilated nitrogen is excreted
- 90% of nitrogen excreted as TAN + 10% as urea (fresh water fish only)
- all TAN is excreted during time period "t"
- non assimilated nitrogen in feces is removed quickly
- no additional mineralization of nitrogenous compounds

The individual assumptions about digestion and ultimate production of ammonia that is diffused across the gill and excreted directly via the feces all lack crispness in their assignment. Thus, we tend to see the rate of ammonia generation as being a "soft" number. For simplicity, one could simply assume 10% of the protein in the feed becomes the ammonia-N generation rate.

Equation 3.9 represents a conservatively high estimate of the P_{TAN} production rate. We use the time period in Eq. 3.9 as one day, while others will use the time period between feedings. In RAS, feed can be fed uniformly over a 24 hour period, thus distributing the ammonia load

uniformly over the entire day as well. If a uniform 24 hour feeding is not used, then the equation should be adjusted and the time period should be the time between feedings or if a single feeding per day is used, then use 4 hours as the time period as an estimate of the time for the ammonia to be excreted from a feeding event. The assumption that all of the TAN is excreted in a finite period of time (t) between feedings is founded in evidence that metabolic activity increases during the hours following feedings (Page and Andrews, 1974; Ruane et al. 1977). Although the value of t is dependent on many biological variables, experience has indicated that fish metabolic activity peaks from 1 to 4 hours following feeding. One quickly will conclude that many smaller feedings evenly spaced during the day would serve to minimize high values of P_{TAN}. In fact, this is a strategy employed in the production of fish in tanks through the use of automatic feeders or demand type feeders.

Carbon Dioxide, CO_2

Carbon dioxide (CO_2) is an important, but largely overlooked water quality limiting parameter. This is probably because until recently, most systems were generally low density (less than 40 kg/m^3) and relied on aeration as the main means of supplying oxygen. This type of management also kept CO_2 values at low levels, e.g., less than 20 mg/L. However, loading rates have increased in recent years, and it became necessary to inject pure oxygen into these systems, instead of using aeration. As result, the natural stripping of CO_2 that occurs when using aeration systems was no longer taking place. We now need to apply other means for CO_2 control.

The generation of CO_2 is based upon chemical stoichiometry by relating CO_2 production to oxygen consumption (1.375 is the ratio of molecular weights of the two gases, 44/32):

CO_2 = 1.375 grams produced for each gram O_2 consumed
(by both fish and bacteria)

Please refer to Chapter 10 for more extensive discussion on carbon dioxide control. Additionally, please note that a computer software program to design CO_2 removal systems is provided in the Appendix. The relationship between CO_2 and alkalinity is shown in Fig. 3.3 for an alkalinity value of 100 mg/L. CO_2 concentration is proportional to alkalinity within normal ranges of pH encountered in aquaculture applications, e.g., pH of 6 to 8.5.

The available data on acceptable levels of CO_2 are difficult to interpret, at best. The authors have maintained tilapia systems in excess

of 100 mg/L on a sustained basis, but this seems to be the exception. Colt and Watten (1988) recommend that the maximum level of CO_2 should not exceed 20 mg/L, while Needham (1988) recommends the maximum not to exceed 10.0 mg/L. Alabaster et al. (1957) report that in well aerated water, CO_2 levels don't become toxic for rainbow trout under 100 mg/L. On the other hand, 10 mg/L caused mortalities at a pH level of 4.5, and mortalities occurred at 20 mg/L CO_2 when the pH was 5.7 (Lloyd and Jordan, 1964). In other research, Piper et al. (1982) state that 40 mg/L CO_2 had little effect on juvenile coho salmon. However, they also mention that CO_2 in excess of 20 mg/L may be harmful to fish, and where DO levels drop to 3–5 mg/L, lower concentrations may be detrimental and long term exposure of one year or more should not exceed 12 mg/L. Smart (1981) argues that fish are able to acclimate to elevated levels of CO_2. High CO_2 levels may lead to nephrocalcinosis, the presence of white calcareous deposits in the kidney. The severity of this condition appears to vary greatly according to diet and environmental factors as well and in particular the form of alkalinity replacement used. Chen et al. (2001) reported that the usage of agricultural grade limestone in place of sodium bicarbonate in an intensive tilapia system led to increased levels of nephrocalcinosis.

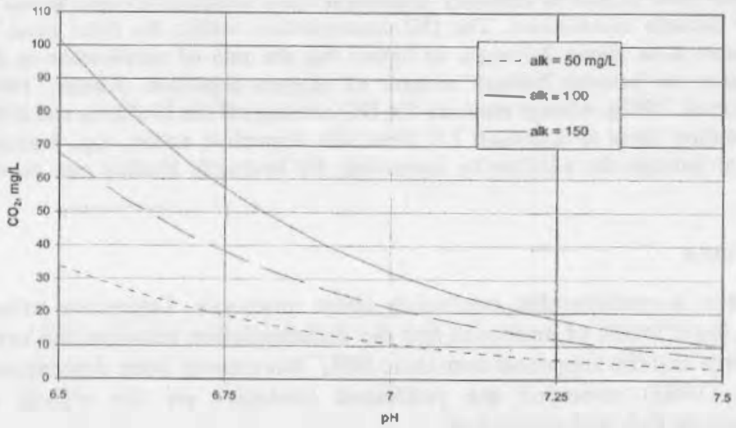


Figure 3.3 Concentration of CO_2 Versus pH at alkalinities of 50, 100 and 150 mg/L (see spreadsheet at web site, www.bee.cornell.edu/aqua for both fresh and saltwater conditions)

A key point in the above discussion is that fish successfully acclimate to slowly changing water quality conditions, such as increases in the level of CO_2, but are adversely affected by sudden changes of water condition. Using CO_2 as an example, fish acclimated to CO_2 levels

of 20 mg/L may die if exposed to a sudden spike to 80 mg/L CO_2 while other fish may exhibit good growth and feed conversion ratios at a gradually created but sustained level of 80 mg/L CO_2. An advantage of RAS is that system water quality can be maintained at fairly uniform values in a normally functioning system, but failures or malfunctions of the system that cause certain water quality parameters to fluctuate widely may cause fish to suffer or die.

SUSPENDED SOLIDS

The effective control of solids generated in RAS is probably the most important task that must be accomplished to ensure long-term successful operation of an RAS. This aspect of RAS is discussed in more detail in Chapter 5, but is also presented here as the suspended solids are a component of water quality. The quantity of suspended solids or Total Suspended Solids, TSS, generated per unit of feed being fed is estimated as:

TSS = 0.25 · kg feed fed (dry matter basis)
(from 20% to 40% of feed fed dry basis)

TSS is treated as a dilute waste. TSS design concentrations in RAS will be in the 10 to 30 mg/L range. Even after concentrating TSS with some type of treatment process, a certain volume of water will still contain only around 0.5 to 1% solids on a dry matter basis. In comparison, cow manure is 20% solids. TSS captured in a settling basin has a fluffy consistency and will require substantial volumetric space depending upon frequency of cleaning. As a "rule of thumb", assume that each kg of dry feed fed will produce approximately 8 liters of liquid waste, i.e., one lb feed produces one gallon of manure.

NITRATE

Nitrate nitrogen (NO_3 is the end product of the nitrification process. In general, high concentrations of nitrate are not extremely adverse to RAS water quality. We have maintained some salmonids systems at nitrate levels above 1,500 mg/L without impact on the fish. Nitrogen should be conserved throughout the nitrification process. Thus, if 1 kg per day of TAN is being produced, then 1 kg of nitrate-N is being produced. The equilibrium concentration of nitrate will therefore be directly dependent upon the overall water exchange rate through the system. An effective exercise is to calculate the steady state nitrate-N balance assuming some water exchange rate through the system (see Eq. 3.4). Nitrate-nitrogen is relatively non-toxic to fish and as such will not influence the controlling flow rates in the system. One can choose some value such as 500 mg/L if you want a number to work with.

3.3 FISH GROWTH

The premise of RAS design is that we are endeavoring to grow fish at some defined rate, and that rate then defines the required fish feeding rate. The fish feeding rate in turn then defines waste generation loads and oxygen consumption. A convenient way of defining fish growth is based upon a temperature unit approach and some defined number of temperature units to create a unit growth rate, e.g., one cm or inch per month

$$Growth = \frac{T - T_{base}}{TU_{base}} \qquad (3.10)$$

Equation 3.10 predicts growth based with units of inches per month. The T_{base} and TU_{base} terms are defined in Table 3.2 and based upon historical observation and analyzing hatchery records. The terms for trout are from Piper et al. (1982) and the tilapia and perch terms are from the author's unpublished data:

Table 3.2 Temperature Growth Units for Trout, Tilapia, Perch, and Hybrid Striped Bass, C° (°F)

	Trout	Tilapia	Perch	H.Striped Bass
T_{base}	0 (32)	18.3 (65)	10 (50)	10 (50)
TU_{base}	6.12 (28)	3.28 (15)	5.47 (25)	5.47 (25)
T_{max}	22.2. (72)	29.5 (85)	23.9 (75)	23.9 (75)

Use Eq. 3.10 subject to the limitations that if T is greater than T_{max}, then calculate the growth at T_{max}. Note that excessive temperatures will compromise growth and/or feed conversion.

Example #1: Consider tilapia and a water temperature of 26.7°C (80°F):

$$Growth\ \frac{cm}{month} = \frac{26.7 - 18.3}{3.28} = 2.54\ \frac{cm}{month}$$

CONDITION FACTOR AND FISH WEIGHT

The weight of fish can be mathematically related to their length by using a term called the condition factor (K or CF); the bigger the condition factor, the more weight per unit length. This concept was first introduced by Fulton (1902) and is expressed quantitatively using Equations 3.11 for either metric or English units. Values for the condition factors and exponent for various fish are listed in Tables 3.3a and 3.3b. Note that the value of n for fish is near 3.0. The Condition Factor is often referred to as the "K factor" or "CF Factor". For a given length, the bigger the K or CF factor, then more the weight of a particular fish. Each fish species will have an associated K or CF factor value to describe expected or normal body condition. The value of this factor is influenced by age of fish, sex, season, stage of maturation, fullness of gut, type of food consumed, amount of fat reserve and degree of muscular development (Barnham & Baxter, 1998).

Table 3.3a Condition factors for various fish for use in Eq. 3.11a (from Piper et al. 1982, this book's authors*, or Blancheton**, 2010)

Species	CF (L inch, Wt, lb)	K (L cm, Wt, g)
Tilapia*	750-900	2.08–2.50
Tilapia < 1 gm*	500	1.39
Rainbow and Brown Trout	400	1.11
Lake Trout (or Sea trout*)	250	0.69
Charr	520	1.45
Hybrid Striped Bass*	720	1.99
Perch	490	1.36
Muskellunge	150	0.42
Northern Pike	200	0.56
Largemouth Bass	450	1.25
Walleye	300	0.83
Grouper* (*Plectropomus pessuliferus*)	550	1.53
Sea bass** ~ 220g (or 135g (*Dicentrarchus labrax*)	680 (867)	1.88 (2.40)

Note: Piper, pg. 406, uses CF's that are 10 times the above values, e.g., Piper would report the trout CF as 4,000 and would divide by 10,000,000 instead of 1,000,000 in Eq. 3.11.

Thus, knowing the K or CF value is a good tool to gauge current feeding protocols for a given tank of fish. A K or CF too high means you are over-feeding the fish and a K or CF too low means you are under-feeding the fish. Using the K or CF from Table 3.3a and the fish length, weight can be calculated:

$$WT(g) = \frac{K*(L_{cm})^n}{10^2} \quad \text{or} \quad WT(lbs) = \frac{CF*(L_{inches})^n}{10^6} \qquad (3.11a)$$

Table 3.3b Condition factors for various fish for use in Eq. 3.11b (from, Huguenin and Colt, 2002)

Species		K x 10^6	n
Atlantic sturgeon	*Acipenser oxyrhynchus*	1.1402	3.18
Channel catfish	*Ictalurus punctatus*	5.160	3.11
Largemouth bass	*Micropterus salmoides*	12.748	3.00
Chinook salmon	*Oncorhynchus tshawytscha*	8.190	3.00
Muskellunge	*Esox masquinongy*	4.429	3.00
Northern pike	*Esox lucius*	5.012	3.00
Rainbow trout	*Oncorhynchus mykiss*	11.224	3.00
Walleye	*Sander vitreus*	8.303	3.00
European eel	*Anguilla anguilla*	0.04302	3.63
Milkfish	*Chanos chanos*	8.989	2.99
Pacific bonito	*Sarda chiliensis*	7.729	3.09
Crayfish	*Procambarus acutus acutus*	8.026	3.32
Freshwater prawn	*Macrobrachium rosenbergii*	1.305	3.42
Red swamp crayfish	*Procambarus clarkia*	8.837	3.28
Shrimp	*Panaeus stylirostis*	15.2	3.10
Shrimp	*Panaeus vannamei*	9.88	3.05
American lobster[1]	*Homarus americanus*	589	3.07
Blue crab[2]	*Callinectes sapidus, female*	287.4	2.64
	Callinectes sapidus, male	181.4	2.78
Rock crab[2]	*Cancer irroratus*	87.10	3.14
Sea Turtle[2]	*Chelonia mydas*	1659	2.54
Squid[3]	*Loligo pealai*	1809	2.15
Ocean quahog	*Arctica islandica*	68.436	2.89

[1]carapace length [2]carapace width [3]dorsal mantle length

Or weight can be calculated using the K and n values from Table 3.3b and using Eq. 3.11b:

$$WT(g) = K*(L_{mm})^n \qquad (3.11b)$$

where length (L) is in mm and weight (WT) in grams. Note the actual value of K is the value of K in the table x 10^{-6}.

Example #2:

Tilapia that are 17.8 cm (7 in) and using a K of 2.08 (CF of 760), Table 3.3a:

Solution

$$WT(g) = \frac{2.08 * (17.8\,cm)^3}{10^2} = 117\ g$$

or

$$WT(lbs) = \frac{760 * (7\,in)^3}{10^6} = 0.26\ lbs$$

WEIGHT GAIN

Now, we can calculate actual weight gain by calculating the weights associated with two specific fish lengths over the time period required to achieve this growth. In the previous example, we grew 17.8 cm (7 inch) tilapia for one month at 26.7°C (80°F), which produced 2.54 cm (1 inch) of growth to a size of 20.3 cm (8 inches). The new weight is then:

$$WT_{New} = \frac{2.08 * (20.32)^3}{10^2} = 175\ g\ or\ 0.38\ lbs$$

The relative efficiency of converting feed energy into animal flesh is described by a term called the feed conversion ratio, FCR, sometimes referred to as feed to gain ratio, or FG. FCR is used to calculate feeding rates required to achieve projected growth rates:

$$FCR = \frac{feed}{gain} \tag{3.12}$$

$$Feed = FCR \cdot gain \tag{3.13}$$

$$\frac{feed}{month} = (WT_{new} - WT_{old}) \cdot FCR \tag{3.14}$$

Returning to our example of tilapia increasing in length for 17.8 cm (7 inch) to 20.3 cm (8 inches) in one month, the feed per fish required per month is:

$$\frac{feed}{month} = (175\ g - 117\ g) \cdot FCR$$

In the above example, each tilapia will gain 58 g (175g – 117g = 58 g) over the one month period. Converting this to a daily rate and using 30.5 days per month, the daily gain is:

$$daily\ gain = \frac{58\ g}{month} \cdot \frac{month}{30.5\ day} = \frac{1.90\,g}{day} \tag{3.15}$$

The expression in Eq. 3.15 is calculating the average daily gain based upon the average length gain over the month. Since weight gain is cubically related to length gain, the gain for the "last-day" of the month can be calculated based upon the length incremental gain on the last-day of the month. This concept is demonstrated in the following example.

Example #3:

Calculate the maximum daily feeding rate for a tank containing 10,000 tilapia at a harvest weight of 0.907 kg (2.00 lb) with a K = 2.08 (CF=760); a tank temperature of 26.7°C (80°F); and a FCR of 1.1.

Solution

First calculate the length of a 0.907 kg tilapia:

$$L(cm) = \left[\frac{100 * 907g}{2.08}\right]^{1/3}$$

$$L\ (cm) = 35.20$$

From Eq. 3.10

$$Growth\ \frac{cm}{month} = \frac{26.7 - 18.3}{3.28} = 2.54\,\frac{cm}{month}$$

Daily Increment of length (cm) = 2.54/30.5 = 0.083 cm

$$WT\,(day-1) = 2.08\ (35.20 - 0.083)^3/10^2$$

$$WT\ gain = 907\ g - 901g$$

WT gain /day/fish = 6.4 g

$$\text{Tank feed/day} = 10{,}000\ \frac{fish}{\text{tank}} * 6.4\ \frac{g\ gain}{fish \cdot day} * 1.1\ \text{FCR}\ \frac{g\ feed}{g\ gain}$$

= 70.4 kg (154 lb) feed per day per tank

Feeding charts can be constructed using the above approach. Obviously, these calculations are well suited for spreadsheet applications.

FCR can be very erratic. Assuming good management and good quality feed (protein content of 38 to 42%); guidelines for FCR are as follows for tilapia:

0.7 to 0.9 (tilapia < 100 gram)
1.2 to 1.3 (tilapia > 100 gram)

Similarly, given length growth, the weight gain for any particular fish species can be calculated by using the condition factor for the particular species (both the tilapia and perch growth constants were developed by the authors).

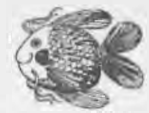
Remember, all the "P" terms in Eq. 3.2 are related to the feeding rate term. Essentially, all design aspects of RAS technology are related to or in fact are based upon the daily design feeding rate. The daily design feeding rate is the critical value needed in designing a complete RAS. It should make some intuitive sense then, that one must calculate the largest anticipated feeding rate in order to properly design the water conditioning components of an RAS. And when will this largest feeding rate occur? The answer is when the fish tank reaches its largest biomass. This is also where many designs fail, since the largest fish mass and largest feeding rate is the most stressful condition on the RAS.

Any of the individual water quality parameters can quickly become the defining variable for water quality control, meaning the water flow rate through a particular water conditioning component is not sufficient to remove the generated pollutant load, e.g., CO_2 removal device. The water parameters always come to an equilibrium based upon a balance between production loads; water flow rate and change in concentration for different water quality variables across the conditioning component (see Eq. 3.6). The challenge to the designer and manager is to make sure these equilibrium values are at least as "good" as the design target values for these variables.

WET WEIGHT MEASUREMENTS

Both terms in the FCR are wet basis, but while feed will typically be from 5 to 10% moisture, the gain term is more nearly 80 to 85% moisture. FCR values for broilers, pigs, and beef are something like 2.0, 2.5 and 3.0, respectively, while in fish; the FCR value may be less than one. Quite typically FCR values will be less than one for fingerlings up to a 100 gram or larger in size. This sometimes confuses people as to how the FCR ratio could be less than one, but it is because a comparison of wet weight to dry weight terms is being made.

3.4 DESIGN EXAMPLES

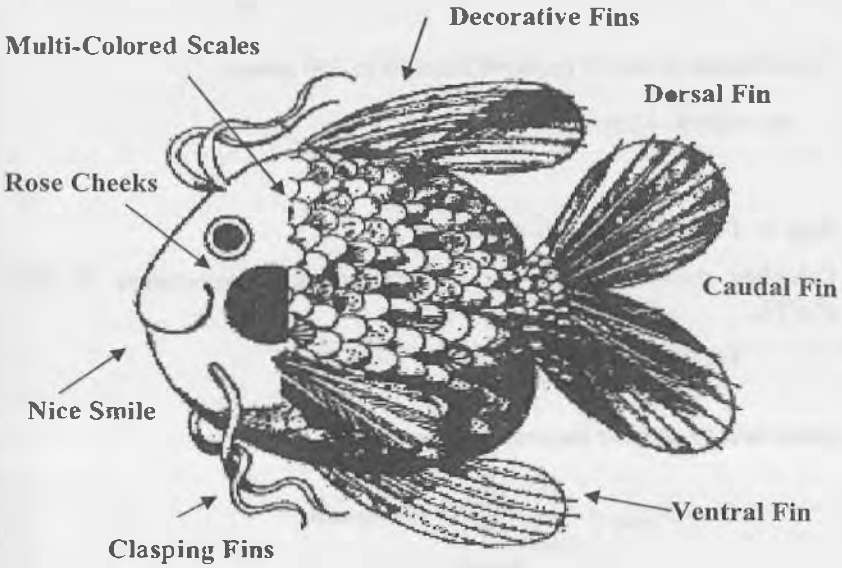


Omega Industries (OI) requires an engineering plan for the construction and operation of an Omega Fish Aquaculture Facility (OFAF). OI's market studies demonstrate a market for 1,000 metric tonnes (live weight) at 750 g (1.65 lbs), whole on ice at $2.50/kg. Several commercial, state and federal hatcheries are producing fingerlings at a mean size of 50 g. For competitive reasons, OI would like to limit its initial efforts to a very small segment of this market to gain experience with intensive recirculating aquaculture systems, hire and train a management and sales team and conduct market surveys to determine markets and opportunities for value added processing. OI is also looking for small scale, local production in urban settings to take advantage of economic development funds, low interest loans and grants. This will also allow OI to market their product as both locally grown, employing a sustainable "green" technology with a small carbon footprint and using local resources and personnel. To this end, only 45 metric tonnes (100,000 lbs) per year are planned for the initial operation with potential for future expansion using the same technology.

Name: Omega Fish (a.k.a. Fakefish, Aquamal and Baloney Fish)

Scientific Name: **Physhi physhy**

Habitat: The Omega fish was originally found in the shallows of large lakes and slow moving rivers away from areas of excessive vegetation. They feed on anything that appears to be organic and moves, is visible, and is smaller than the opening of their mouths. The Omega fish can be naturally found on one South Pacific Island just south of Bali Hai. They have, however, been transplanted so that they are found throughout the world. Naturally bad tempered, but very delicious, and a perfect fish for RAS culture.



Fish Growth: A convenient way of defining fish growth is based upon a temperature unit approach and some defined number of temperature units to create a unit growth rate, e.g., cm or inches per month

$$Growth = \frac{T - T_{base}}{TU_{base}}$$

The above equation predicts growth in units of cm or inches per month. It turns out that like most fish, Omega Fish grow in length very uniformly over the production cycle. Thus, it is easy to estimate time required for growout, given the total change in length of the fish. The T_{base} and TU_{base} terms are defined in Table 3.2 and are based upon historical observation and analyzing hatchery records. If the Omega Fish are reared under good water quality conditions and a water temperature of 28°C (82°F), the predicted growth rate is:

$$Growth \frac{cm}{month} = \frac{28 - 18.3}{3.28} = 3.0 \frac{cm}{month} \qquad (1.16 \frac{in}{month})$$

Feed Rate: It has been determined by the feed manufacturer that the normal growth requirements for the Omega Fish can best be met by feeding the fish the following amounts:

$$Feed_{rate} = 0.020 * Fish_{weight} * \frac{15cm}{Fish_{length}}$$

The Omega Fish food can be obtained from the Omega Fish Food Manufacturing Company of St. Louis, Missouri, founded in 1948 and still a family owned enterprise. It should be noted that this kind of information is not always available for all species, but that an estimate can be made based on the feed conversion ratio (FCR) which can be estimated based on prototype production trials.

Length/Weight Relationship:

$$Fish_{weight} = \frac{K * (L)^3}{100}$$

Where:

$Fish_{weight}$ = fish weight, grams; L = length, cm
K = conditioning factor and ranges between 2.0 to 2.4

Fish Stocking Density: One of the first and most important questions that must be addressed in the design is the mass of fish that can be stocked in the tank. The number of fish and their individual weight will define the feeding rates from which all other individual engineering components are designed. The mass of fish that can be stocked per unit volume ($D_{density}$) will depend on both the fish species and the fish size (and the risk level the producer is willing to accept). What is presented here is an approach that is based upon body length (L) to estimate the number of fish that can be carried per unit volume of tank:

$$D_{density} = \frac{L}{C_{density}}$$

where:

$D_{density}$	Density in kg/m^3 (lbs/ft^3)
L	Length of fish in cm (inches)
$C_{density}$	Omega Fish: 0.24 for L in cm (1.5 for L in inches)

These values were developed based upon experience originally with brook trout and were later adapted to Omega Fish. There is a tendency to over-stock smaller fish. Our experience indicates that Omega Fish can be stocked in much higher numbers, while the animals are small. However, crowding is risky, as it can have severe consequences on small animals during their rapid growth stage, e.g., fish less than 13 cm (5 inches) in length. Visual observations can be misleading, since you will see only the active dominant fish. Sampling fish populations will be your best guide to whether or not the stocking densities are too high.

The above criteria have been the subject of 31 doctoral dissertations, master theses, and $7,500,000 worth of research grants from Federal, State, RAC's, NGO's, for-profits and non-profits (although they were initially planned to be for-profits).

The Omega Fish is the construct of Dr. Ronald D. Mayo, Executive Vice President, Kramer, Chin & Mayo, Inc. The Omega Fish was introduced in February, 1974 in Technical Reprint No. 30, A Format for Planning a Commercial Model Aquaculture Facility, presented at Northeast Fish and Wildlife Conference, February 25-28, 1974, Great Gorge, New Jersey.

PRODUCTION STRATEGY AND CIRCULATION REQUIREMENTS

Assume a target production goal of Omega Fish of 100,000 lbs (45.5 tonnes) per year with a target market size fish of 750 g (1.65 lbs), whole on ice. Fingerlings are purchased from a biosecure facility at a mean size of 50 g. Example *assumes no mortality* or culling between stages.

Step 1: Calculate change in length from fingerling stocking to harvest

The following calculation is employed to determine the growth period of Omega Fish from 50 grams to 750 grams, at 28°C (83°F); assume a K = 2.10 (CF = 760).

Calculate length of fingerlings at 50 and 750 grams:

$$L\,(cm\ at\ 50\,g) = \left[\frac{100\cdot 50}{2.10}\right]^{1/3} = 13.4 \text{ cm}$$

$$L\,(cm\ at\ 750\,g) = \left[\frac{100\cdot 750}{2.10}\right]^{1/3} = 32.9 \text{ cm}$$

Total change in length required from 50 to 750 grams:

$$\Delta L = (32.9 - 13.4) = 19.5 \text{ cm}$$

Step 2: Determine time for growout

Calculate growth rate at the specified rearing temperature of 28°C (83°F):

$$Growth_{rate} = \frac{28-18.3}{3.28} = 2.96\,\frac{cm}{month}$$

Total time to achieve the required length:

$$Growth_{period} = \frac{19.5\,cm}{2.96\,\dfrac{cm}{month}} = 6.6\ months$$

Step 3: Determine production strategy

Note that this growth could occur in a single tank and the fish remain there for 6.6 months, or the growth could be managed to occur in three tanks that are managed sequentially. If a three stage scheme is used with equal time at each state, then the fish would reside in each tank for 1/3 of the total growth time, or

Time in a particular tank = 6.6 months/3 size classes
= 2.2 months/size class
(or 9.5 weeks in each tank)

A very simple concept here that is sometimes missed is that whatever the production cycle is for a stage will determine how many tanks are needed for that stage. If 10 weeks are required for the final production stage, and weekly harvests are required, then 10 final growout tanks will be required. If fish are harvest every two weeks, then only 5 final growout tanks are needed. The other 20 weeks of the growout cycle can be distributed however you choose, but their cumulative growth time must add up to 20 weeks. More commonly, the time the fish remain in the third stage might be increased (e.g. stage 1 for 5 weeks, stage 2 for 10 weeks, and stage 3 for 15 weeks), so there is less stress on sorting and moving fish from stage 2 to stage 3 (this scheme allows you to sort and move smaller fish which is always easier on you and the fish).

To keep things simple in this example, assume that the Omega Fish will be harvested weekly and kept in each stage an identical time. This will require designing a fingerling to juvenile to growout production strategy that requires 10 tanks (rounding the 9.5 weeks to 10 weeks) per stage of increasing size, plus systems for circulation, solids removal, biofiltration, and gas exchange. During each stage, the change in length of the fish would be the total increase (19.5 cm) divided by the number of stages (3) or 6.5 cm. Using the length/weight relationship, the initial and final weights can be calculated at each stage as shown in the Table 3.4.

Table 3.4 Initial and Final Weights and Lengths of the Three Stage Omega Fish Production Strategy

	Initial Wt Size	Final Wt Size	Final Tank Biomass kg	Feed Rate (%bw/day)	Final Feed Rate
Fry/Juvenile:	50 g 13.4 cm	165 g 19.9 cm	193 kg	1.56% (1.1FCR)	3.0 kg
Fingerling:	165 g 19.9 cm	386 g 26.4 cm	450 kg	1.28% (1.2 FCR)	5.7 kg
Growout:	386 g 26.4 cm	750 g 32.9 cm	875 kg	1.11% (1.3 FCR)	9.6 kg

Step 4: Calculate weekly harvest weight

Assume that 52 weekly harvests are conducted:

$$Harvest_{weekly} = \frac{100{,}000\ lbs/year}{52\dfrac{weeks}{year}} = 1{,}925\ lbs\ per\ week$$

or:

$$Harvest_{weekly} = \frac{45{,}500\ kgs/year}{52\dfrac{weeks}{year}} = 875\ kgs\ per\ week$$

Step 5: Determine the number of fish per tank at harvest:

Note no allowance was made for mortalities, but they are a part of life and are best estimated based on actual production experience.

$$Fish_{Tank} = \frac{875\ kg/tank}{750\dfrac{g}{fish}} * \frac{1000\ g}{1\ kg} = 1{,}167\ fish\ per\ Tank$$

Step 6: Estimate final tank biomass at each stage

The final biomass in each of the three production tanks will be the final weight times the number of fish.

$$Biomass_{Tank} = 1{,}167\ fish\ per\ Tank * Weight_{Final}$$

These are also summarized in Table 3.4.

Step 7: Estimate final feed rate at each stage

Often the daily feed rate as a percent of the fish weight (% bw) is provided by the feed manufacturer or other sources. If this data is not available, then an estimate of the final feed rate at each stage can be made by calculating the weight of the fish the day before harvest, subtracting that from the weight at harvest and adjusting with an estimated feed conversion rate (FCR). As an example, the final weight for the growout stage is 750 g with a corresponding length of 32.93 cm. The Omega Fish growth rate per day is 0.0932 cm/day. Thus the length one day before harvest is 32.809 cm and converting this by using the condition factor into weight, yields a weight of 743.65 g. Thus the increase in weight per fish during the final day of growout is 6.35 g multiplying times the number of fish (1167), yields a total increase in fish weight for the entire tank of fish of 7.41 kg.

The feed conversion rate (FCR) is the ratio of feed to weight gain and varies with age, feed characteristics, and most importantly, management skills. Assuming a FCR of 1.3 for the growout stage, yields a final feed rate of 9.64 kg or 1.11 percent body weight per day. A similar calculation can be carried out for the other two production stages, juvenile and fingerling, and also are summarized in Table 3.4. This is a critical calculation since all of the other engineering design values are based on this feed rate, including circulation requirements, solids capture, biofiltration, aeration/oxygenation, gas stripping and disinfection.

Step 8: Determine the controlling flow rate for this design problem (i.e., dissolved oxygen, ammonia-nitrogen removal, carbon dioxide stripping, or Total Suspended Solids)

In Step 8 we will determine the flow rates to control each of the design variables. These calculations are always based upon some design feeding rate for a tank or more typically some collection of tanks that are placed on a common life support system (biofilter, gas transfer devices, solids removal units, etc). In the example below, we do all these calculations as if there were only one fish tank and that it reaches a maximum feeding level of 9.6 kg/day. We do this at this point to keep the problem as simple as possible and to create the least amount of confusion. In later chapters, we will present the more common approach of designing for feeding loads associated with a LSS that supports a group of tanks that are probably at different levels of maturity so that daily feeding loads become as constant as possible, as opposed to a single tank which slowly climbs to its maximum feeding load and then you harvest (or move) the fish.

Water circulation is the mechanism by which oxygen is transported into a fish culture vessel and the waste products being generated within are removed. The design of a recirculating aquaculture system (RAS) should insure that the important parameters affecting water quality and fish productivity, e.g. oxygen, ammonia-nitrogen, carbon dioxide, and suspended solids (TSS) are properly balanced. This requires calculating the required flow rates via mass balance equations to maintain the design water quality variables at or below (above in the case of oxygen) their maximum tolerable or design target values. Then the system must be operated at the highest flow rate calculated for these four critical water quality parameters. Obviously, the maximum flow rate used to maintain the constraining water quality parameter will be higher than necessary for the others, which simply means these water quality parameters will be at "better" values than design targets. The same mass balance approach can be utilized on any variable affecting water quality.

As an example, calculate the required design flow rate for a 100% recirculating flow for the growout tank assuming a feeding rate of 9.6 kg feed/day @ 35% protein (maximum value for the growout stage given in Table 3.4). To accomplish this, we must first calculate the required flow rate for each water quality parameter (oxygen, ammonia, carbon dioxide and TSS) and then identify the controlling parameter.

Compute the required steady state flow rate for maintaining the following water quality levels:

- 5 mg/L O_2
- 2 mg/L TAN
- 20 mg/L CO_2
- 10 mg/L TSS

Assume the following efficiencies for the treatment devices:

- 90% for O_2 transfer
- 35% for TAN removal
- 60% for CO_2 stripping
- 90% for TSS removal

A good place to start is with the General Mass Balance Eq. 3.5, where C_1 is outflow and C_2 is inflow from the fish culture tank and P is the Production rate or consumption (negative) rate:

$$QC_2 + P = QC_1,$$

or $$Q(C_2 - C_1) = -P$$

or $$Q(C_1 - C_2) = P$$

and the impact of a treatment device on the discharge water quality concentration can be calculated as follows:

$$C_{out} = C_{in} + T/100 \times (C_{best} - C_{in})$$

where, C_{in} and C_{out} the water quality concentration entering and leaving the treatment device, T is the treatment efficiency (%) and C_{best} is the absolute best result obtainable by a treatment system, e.g., zero ammonia or saturated oxygen, zero suspended solids. Note that if the device is an oxygen addition unit, the C_{best} term can be increased above atmospheric concentration values for oxygen by increasing the partial pressure above atmospheric oxygen partial pressure in the device. For example, a pure oxygen device will have a C_{best} value of roughly five times the C_{best} value obtained if normal air were used at atmospheric pressure, e.g., trickling tower.

Required Design Flow Rate for Dissolved Oxygen

The major reason most fish die is from lack of oxygen due to a loss of water flow. This is because oxygen is consumed at a fairly high rate

by both the fish metabolism and the biofilter demands. Due to low inherent concentrations of oxygen, "high" flows are required to transport the required oxygen. Flows required to maintain a satisfactory oxygen level are generally the controlling flow rate parameter.

First calculate the effluent dissolved oxygen concentration, C_{out}, using a Speece cone with 90% oxygen transfer efficiency (TE) and a production tank DO level (target value for minimum) of C_{in} = 5 mg/L, and an enriched oxygen stream at 16.0 mg/L from a VSA or PSA generator (Note: Chapter 10 Gas Transfer reviews the calculations to determine oxygen saturation as affected by environmental conditions and gas purity; C_{best} could be as high as 100 mg/L using pure oxygen and a 30 psi environment).

$$C_{out} = C_{in} + T/100 \, x \, (C_{best} - C_{in})$$

Solve for C_{out}, and you obtain:

$$C_{out} = 5.0 \ mg/L + 0.90 * (16.0 \ mg/L - 5.0 \ mg/L)$$
$$C_{out} = 15.0 \ mg/L$$

Second, calculate the oxygen production (P) or in this case consumption (–P) equal to the sum of fish metabolism and bacterial oxygen consumption (heterotrophic and autotrophic)[c]:

$$P = \frac{0.25 \, kg \, O_2 \, by \, Fish}{kg \, feed} + \frac{0.25 \, kg \, O_2 \, by \, Bacteria}{kg \, feed} = \frac{0.50 \, kg \, O_2}{kg \, feed}$$

$$P = \left(\frac{9.6 \, kg \, feed}{day}\right)\left(\frac{0.50 \, kg \, O_2}{kg \, feed}\right)\left(\frac{10^6 \, mg}{kg}\right) = \frac{4{,}800{,}000 \, mg \, O_2}{day}$$

Returning to the General Mass Balance for a RAS:

$$Q_1 C_2 + P = + Q_1 C_1$$

$$Q_1 * (15.0 \text{ mg/L}) + (-4{,}800{,}000) \text{ mg/day} = Q_1 * (5.0 \text{ mg/L})$$

---

[c] Trickling, RBC, and moving bed biofilters are self aerating and in these cases the oxygen load from the bacteria can be set to zero; to be conservative, often some fraction of this bacterial load is still left in the calculation.

$$Q = \frac{\dfrac{4{,}800{,}000 \, mg \, O_2}{day}}{(15.0 - 5.0)\dfrac{mg \, O_2}{L}} = \frac{480{,}000 \, L}{day} \quad or \ 337 \ Lpm$$

Finally, approximately 337 Lpm (89 gpm) of influent tank water at 15.0 mg/L DO is required to satisfy the oxygen demand of the 9.6 kg of feed per day. Similar calculations are used for the other two production units, juvenile and fingerling.

Required Design Flow Rate Total Ammonia-nitrogen

First calculate the effluent concentration, C_{out}, using a non-self aerating biofilter with a 35% removal efficiency (TE) and a production tank TAN level (target value for minimum) of C_{in} = 2 mg/L and a C_{best} value of 0.0 mg/L.

$$C_{out} = 2.0 \ mg/L + 0.35 \ (0 \ mg/L - 2.0 \ mg/L)$$
$$C_{out} = 1.3 \ mg/L$$

The total ammonia-nitrogen (TAN) production (+P) is based on the feed rate and feed protein level (35%).

$$P = \left(\frac{9.6 \, kg \, feed}{day}\right)\left(\frac{0.35 \, kg \text{ Protein}}{kg \, feed}\right)\left(\frac{0.092 \, kg \, TAN}{kg \text{ Protein}}\right)\left(\frac{10^6 \, mg}{kg}\right)$$
$$= \frac{309{,}000 \, mg \, TAN}{day}$$

Returning to the General Mass Balance for a RAS:

$$Q * (1.3 \ mg/L) - 309{,}000 \ mg/day = Q * (2 \ mg/L)$$

$$Q = \frac{\dfrac{309{,}000 \, mg \, TAN}{day}}{(2 - 1.3)\dfrac{mg \, TAN}{L}} = \frac{441{,}000 \, L}{day} \quad or \ 310 \ Lpm$$

Thus, approximately 310 Lpm (82 gpm) of influent water at 1.3 mg/L TAN is required to dilute the tank TAN concentration to 2.0 mg/L from the 9.6 kg of feed per day.

Required Design Flow Rate for Dissolved Carbon Dioxide

Next calculate the required flow to strip the CO_2 production based on a maximum tank concentration of 20 mg/L CO_2, a stripping efficiency of 70% (TE), and a C_{best} value of 0.5 mg/L assuming atmospheric air is used with 320 ppm concentration of CO_2 for stripping.

$$C_{out} = 20\ mg/L + 0.70\ (0.5\ mg/L - 20\ mg/L)$$
$$C_{out} = 6.35\ mg/L$$

Carbon dioxide production (+P) is based on oxygen consumption, where 1.375 kg of CO_2 is produced for every kg of O_2 consumed.

Thus:

$$P = \left(\frac{9.6\ kg\ feed}{day}\right)\left(\frac{0.50\ kg\ O_2}{kg\ feed}\right)\left(\frac{1.375\ kg\ CO_2}{kg\ O_2 consumed}\right)\left(\frac{10^6\ mg}{kg}\right)$$

$$P = \frac{6,620,000\ mg\ CO_2}{day}$$

Returning to the General Mass Balance for a RAS:

$$Q^*\ (6.35\ mg/L) - 6,620,000\ mg/day = Q^*\ (20\ mg/L)$$

$$Q = \frac{\dfrac{6,620,000\ mg\ CO_2}{day}}{(20-6.35)\dfrac{mg\ CO_2}{L}} = \frac{482,000\ L}{day}\ or\ 333\ Lpm$$

Thus, approximately 333 Lpm (88 gpm) of influent water at 12.4 mg/L CO_2 is required to dilute the carbon dioxide concentration to 20 mg/L from the 9.6 kg of feed per day.

Required Design Flow Rate Total Suspended Solids

Finally examine total suspended solids, since this is directly based on feed fed, calculate the required flow to strip the suspended solids production based on a tank concentration of 10 mg/L, a stripping efficiency of 75% (TE), and a C_{best} value of 0.0 mg/L.

$$C_{out} = 10\ mg/L + 0.90\ (0\ mg/L - 10\ mg/L)$$
$$C_{out} = 1.0\ mg/L$$

TSS production (+P) is based on feeding rate (assume feed has 0% moisture content):

$$P = \left(\frac{9.6 kg\ feed}{day}\right)\left(\frac{0.25 kg\ TSS}{kg\ feed}\right)\left(\frac{10^6\ mg}{kg}\right) = \frac{2,400,000 mg TSS}{day}$$

Returning to the General Mass Balance for a RAS:

$$Q\ ^*(1.0\ mg/L) - 2,400,000\ mg/day = Q\ ^*(10\ mg/L)$$

$$Q = \frac{\dfrac{2,400,000\ mg\ TSS}{day}}{(10-1.0)\dfrac{mg\ TSS}{L}} = \frac{267,000\ L}{day}\ or\ 189\ Lpm$$

Thus, approximately 189 Lpm (50 gpm) of influent water at 1.0 mg/L TSS is required to dilute the Total Suspended Solids concentration to 10 mg/L from the 9.6 kg of feed per day.

Table 3.5 summarizes the required flow rates for the three production stages and the ***tank exchange rate*** recommended for proper tank hydraulics and maximum water quality. It is important to note, that the designer/manager must choose the design or target operating conditions to calculate the mass balances. This is why a decision was made to enrich the influent oxygen concentration to 16.0 mg/L in order to reduce the required flow rate for oxygen. These are the "C" water concentration values used in the mass balance equation. These design numbers are species dependent and are continually being refined for RAS applications. Calculating the minimum flows required to maintain targeted values for water quality (and then using the largest minimum value found for all the different water quality variables) will show how sensitive the calculated flow rates are to the value selected for the design value. A typical scenario is to select a value, do the calculations, realize that economics prevents such a high flow rate, and then start to make adjustments in the targeted values, e.g., increase oxygen availability with an enrichment device or using pure oxygen supplementation. In the end, one must choose realistic values at the beginning of the design process and then stay with these choices and the ramifications of the resulting flows required to maintain the mass balances. Do NOT ever compromise on the required flow rates.

Table 3.5 Summary Required Flow Rates for Oxygen, TAN, Carbon Dioxide, TSS and Tank Exchange (*controlling flow rate).

Water Quality Parameter	Juvenile/Fry	Fingerling	Growout
Oxygen*	321 Lpm (85 gpm)	360 Lpm (95 gpm)	337 Lpm (89 gpm)
TAN	95 Lpm (25 gpm)	185 Lpm (49 gpm)	310 Lpm (82 gpm)
Carbon Dioxide	121 Lpm (32 gpm)	208 Lpm (55gpm)	333 Lpm (88 gpm)
TSS	68 Lpm (18 gpm)	132 Lpm (35 gpm)	189 Lpm (50 gpm)
Tank Exchange	321 Lpm (85 gpm) (20 min HRT)	374 Lpm (99 gpm) (30 min HRT)	390 Lpm (103 gpm) (45 min HRT)

A footnote to the above is that you must also calculate the required hydraulic loading for the biofilter you are using as in $m^3/hr/m^2$ (gpm/ft^2). This is because some biofilters will have some required hydraulic loading minimum in order to function properly. This may become the controlling flow rate over and above the flow rates required to maintain water quality conditions. Just remember that if such were the case, it simply means that the equilibrium concentrations for all the water quality parameters would be just a little *better* and the fish will never complain about that!

The final footnote to Table 3.5 is to remember that everything was ASSUMED! In the real world, there are always numerous options to each treatment system that will have higher or lower operating efficiencies than the ones used in this example. The remaining chapters of this book reviews each of these unit operations and hopefully will help the reader to decide which is best suited for their production concept and species.

3.5 References

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LIST OF SYMBOLS

C_x	Concentration of a particular water quality parameter X, $\frac{kg\ of\ X}{10^6\ kg\ water}$, which is same as mg/L
C_0, C_1 and C_2	Concentrations of parameter X crossing the control volume, mg/L
CF	Condition factor that relates fish weight and length, Eq. 3.11 using English units, dimensionless
D_{fish}	Fish density in culture vessel, kg/m^3
F	Feeding rate, kg/day
$F_{\%}$	Feeding rate per day as a percentage of fish body mass, %
FCR	Feed to gain ratio, dimensionless
K	Condition factor used in Eq. 3.11, using metric units, dimensionless
L	Loading rate, kg/m^3/hr
L_{oxygen}	Allowable fish biomass loading due to oxygen availability, kg/ Lpm
PC	Protein concentration or feed, decimal
P_{Oxygen}	Production rate (negative as related to feed consumption) of oxygen, kg/day
P_{CO2}	Production rate of carbon dioxide, kg/day
P_{TAN}	Production rate of total ammonia nitrogen, kg/day
$P_{Solids,\ TSS}$	Production rate of total suspended solids, kg/day
Q_0	Flow rate passing through culture tank (discharge), m^3/day (as kg/day)
Q_1	Water that is recirculated, kg/day

R	Number of water exchanges per hour through the rearing unit, hr^{-1}
t	Time period for feeding daily ration, day
TAN	Total Ammonia Nitrogen, kg
TSS	Total Suspended Solids, kg
T_{base}	Equivalent to the temperature at which no growth occurs, °F (°C)
T_{eff}	Treatment efficiency (removal or addition), %
TU_{base}	Temperature units required to produce an increment of growth per unit time, °F (°C)

CHAPTER 4

CULTURE UNITS[1]

4.0 INTRODUCTION

About the only thing that aquacultural engineers can *"unanimously"* agree upon is that a body of water is required to raise fish. How this body of water should be contained is open to considerable debate. Should the fish be reared in ponds, raceways, or tanks? If tanks are chosen, to what geometry should the tank conform? How deep should a tank be or more descriptively, what should the tank diameter to depth ratio be? And there are numerous other issues. First of all, this book is restricted to tank culture and Recirculating Aquaculture Systems (RAS) technology. Therefore only tank culture is addressed, not open ponds. Within that chosen focus, the paramount issue in all tank culture design is to maximize the capability of the tank system for self-cleaning. With few exceptions, the dominant reason RAS's fail is their inability to self-clean. Outside of the occasional heterotrophic or ODAS (organic detrital algae soup) based systems used for tilapia or shrimp, such as those described by Serfling (2006), recirculating systems that do not effectively and quickly remove fish fecal matter, uneaten feed and other solids from the culture water will never produce fish economically. With poor solids removal, all other individual components of the system will fail to perform efficiently. Thus, the first question that should be asked is whether or not the culture tank will effectively remove solids. The next question that must be addressed is the capability to effectively manage the fish within the culture tank.

These and other issues will be addressed in this chapter. And for the stubborn, raceway design will also be reviewed, briefly, as well as a newly designed hybrid of raceway and round tank design, i.e., mixed-cell raceways.

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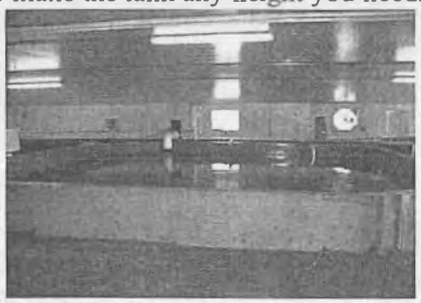
4.1 CULTURE TANKS

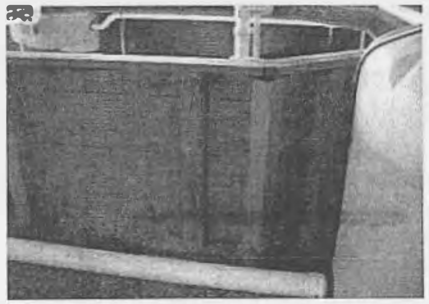
Over the years, the authors have seen almost anything that holds water used as an aquaculture production tank. Polyethylene tanks are used by many small and large scale operations for fry and fingerling production and have gained in popularity due to their low cost and ease of shipment. Their smooth surface makes for easy cleaning and their light weight allows for quick set-up and relocation. They work well for the most part, but because they are very soft and malleable, they need to be well support on the bottom and sometimes on the sides. Another significant problem is the difficulty in making water tight connections through the side walls and bottom. Expensive bulk head fittings are commonly used, but a new product called a Uniseals® works well with polyethylene and is available in sizes from ½ inch to 6 inches (12 to 150 mm). They are rated up to 40 psi and have a 25 year warranty.

The most common material used for aquaculture tank construction is fiberglass. Fiberglass tanks can be manufactured in almost any shape and size. Fiberglass is an incredibly flexible construction material that is easy to cut, drill, and make connections to. Repairs are easy and modifications are simple. If the tank is too tall, cut out a horizontal section and fiberglass the two halves together to make the tank any height you need.



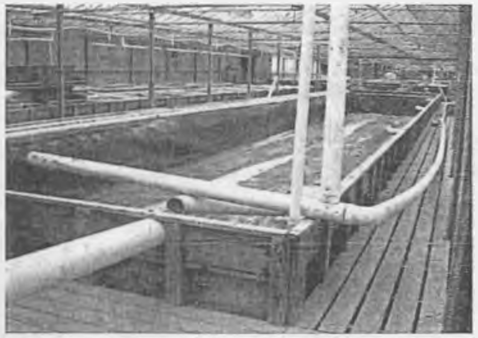
Large fiberglass tanks come in easily transported modules that are field assembled to almost any diameter.

Another simple and inexpensive building material is wood tanks that are lined with a HDPE (High Density Polyethylene) or butyl rubber liner. One author constructed fingerling tanks out of sheets of ¼" plywood panels formed into a circle and then lined with either a swimming pool liner or a more expensive industrial liner (EPDM). The bottom of the tank was lined with several inches of sand into which a central drain line was buried with an outside standpipe. Stainless steel binding straps encircled the tank to insure rigidity. Covering the sand first with a 2 cm board of polyethylene insulation provides thermal resistance and a smooth bottom surface that can be walked on without creating depressions that tend to collect solids.



Another technique is to stack 2x10 or 2x12 and brace them with either 4x4 or galvanized pipe driven into the ground. Add a liner and you have a relatively inexpensive tank. Drain lines can be installed by using either a bulkhead fitting or a flange fitting and a thin sheet of PVC or Plexiglas to secure the liner to the flat side.

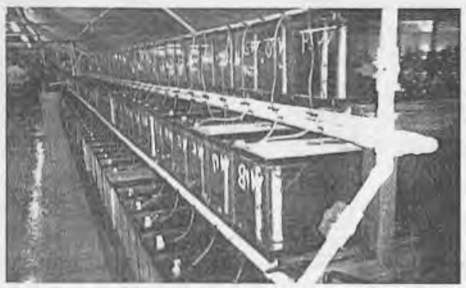
For more permanent tanks, galvanized or epoxy coated steel modules can be bolted together to form extremely large 32 m (105 ft) and deep tanks 4.3 m (14 ft). The bottoms are usually poured concrete and sometimes contain heating coils into which the panels are embedded. Tanks can be partially buried to conserve heat and make fish observations easier.



Another simple method to construct long, shallow raceways is by using plywood panels and a liner. Construction is fast and inexpensive. One of the authors constructed a raceway in an exiting greenhouse using 4x8 plywood panels using standard framing techniques with 2x6's and then bolted the panels together. Another way is to secure 2x10 lumber with 2 inch galvanized pipe driven into the ground, plus a liner. These are usually partially buried to add support to the walls and provide easy access to the fish.

Concrete raceways and tanks have been used for years by Fish & Wildlife agencies to raise sport fish and by some trout and bass producers. They work great, just be sure you plan on using them for generations, because once you build them, there is no going back.

The authors have seen hundreds of abandoned raceways, because there is insufficient water available to operate them. There are some alternatives available, such as cross-flow and mixed-cell raceway designs that do make these systems viable and we may see a comeback in the future.



In planning an aquaculture facility, don't forget the tried and true methods such as simple glass aquarium tanks. Catfish farmers receive $1.40 per kg pond side for their fish; tropical fish growers get 60 cents or more per 1 gram fish ($272.00 per kg)!!

4.2 Stocking Density

One of the first and most important questions that must be addressed in designing a RAS is the mass of fish that can be stocked in the tank. The number of fish and their individual weight will define the feeding rates from which all other individual engineering components are designed. The mass of fish that can be stocked per unit volume ($D_{density}$) will depend on both the fish species and the fish size. What is presented here is an approach that is based upon body length (L) to estimate the number of fish that can be carried per unit volume of tank:

$$D_{density} = \frac{L}{C_{density}} \qquad (4.1^a)$$

where:

$D_{density}$	Density in kg/m^3 (lbs/ft^3)
L	Length of fish in cm (inches)
$C_{density}$	tilapia: 0.24 for L in cm (1.5 for L in inches)
	trout: 0.32 (2.0)
	perch: 0.40 (2.5)
	hybrid striped bass: 0.45 (2.8)

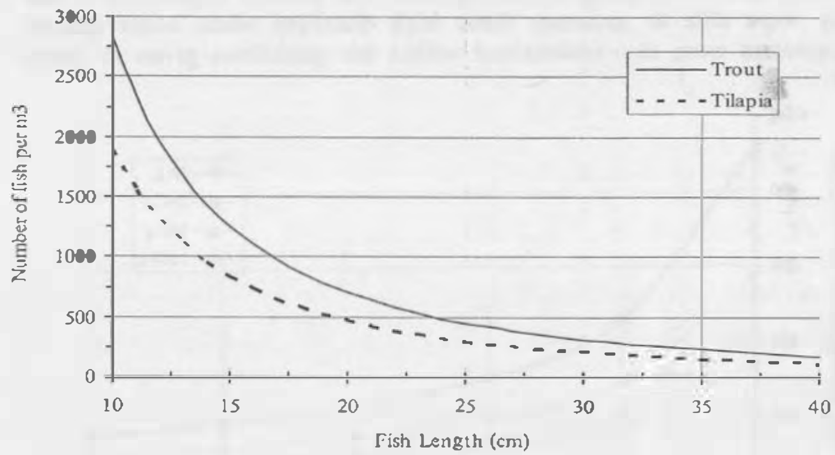


Figure 4.1 Fish per m^3 as a function of body length for Trout (CF= 1.11, C=0.32) and Tilapia (CF= 2.40, C=0.24).

[a] Symbols are defined at the end of the Chapter.

These values were developed based upon experience originally with brook trout and were later adapted to tilapia. There is a tendency to overstock smaller fish. The authors experience indicates that tilapia can be stocked in much higher numbers while the animals are small. However, crowding is risky, as it can have severe consequences on small animals during their rapid growth stage, e.g., fish less than 13 cm (5 inches) in length. Visual observations can be misleading, since you will see only the active dominant fish. Sampling fish populations will be your best guide to whether or not your stocking densities are too high.

Good management should produce a population that has a standard deviation that is less than 10% of the population mean (standard deviation divided by the mean is defined as the coefficient of variation and is generally presented as a percentage). If this value increases, it is indicative of over-stocking, under feeding, or other management problems. The density relationship given above and expressed in Fig 4.1 and Table 4.1 also show what might be considered excessive stocking densities for the larger fish. For example, trout cultured at the higher densities would probably develop eroded fins. However, wide variations in population size may be more related to poor water quality and less than optimum effectiveness of feed distribution than the stocking density. For example, if you use a sinking feed you may be able to stock more heavily than when using a floating feed. The authors experience is that they were able to maintain these high densities when water quality parameters were also maintained within the guidelines given in Table 3.1.

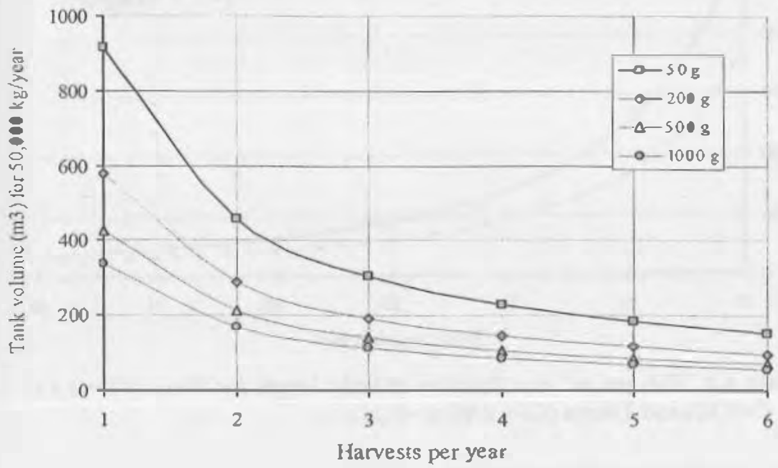


Figure 4.2 Required volume in m^3 to grow 50,000 kg of Tilapia as affected by fish size and number of harvests per year.

Table 4.1 Stocking Densities for Tilapia and Trout as a Function of Body Length

	Length (inch)	Weight (g)	Number/Volume (fish/gal)	Mass/Volume (lbs/gal)
English Units: Tilapia: C=1.5, CF=800: Trout: C=2, CF=400				
Tilapia	1	0.4	111	0.09
	2	3	28	0.18
	4	23	7	0.36
	6	78	3.1	0.53
	8	186	1.7	0.71
	10	363	1.1	0.89
	12	628	0.8	1.07
	14	997	0.6	1.25
Trout	1	0.2	167	0.07
	2	1.5	42	0.13
	4	12	10	0.27
	6	39	4.6	0.40
	8	93	2.6	0.53
	10	182	1.7	0.67
	12	314	1.2	0.80
	14	498	0.9	0.94
SI Units: Tilapia: C=0.24, K=2.08: Trout: C=0..32, K=1.11				
	(cm)	(g)	fish/m^3	kg/m^3
Tilapia	2	0.2	47,000	8.4
	4	1.4	11,800	17
	8	11	2,940	34
	12	38	1,300	50
	15	75	840	63
	20	83	470	83
	25	346	300	105
	30	599	210	126
	35	951	155	147
Trout	2	0.1	70,510	6.3
	4	0.7	17,627	13
	8	6	4,407	25
	12	19	1,959	38
	15	37	1,254	47
	20	89	705	63
	25	173	451	79
	30	299	313	95
	35	475	230	110

The relationship between required tank volume, fish harvest size, and the harvests per year (turnovers) are shown in Fig. 4.2 for tilapia. It can be seen from this figure, that by increasing the number of harvests or turnovers per tank, the volume required for the production of a given amount of biomass is significantly reduced. This is easily realized by splitting the growout period into several stages

The volume required for production of the fingerlings as a percentage of the final volume required for the market size fish (assuming the same number of animals) is shown in Fig. 4.3. This figure can be used as a quick guideline on tank sizes needed for fingerling production. For example, if you intended to sell 1,000 gram tilapia, the tank volume required for 100 g tilapia would be 21% of the volume required for the 1,000 g market fish. In equation form, this relationship can be expressed as:

$$Y = 0.0100W^{0.6667} \tag{4.2}$$

Equation 4.2 is for a "standardized" fish of 1,000 weight units at maturity. For a particular growth scenario, substitute the particular size fish as a percentage of its mature size and 1,000 to standardize the size. For example, a 200 g fish on a 500 g target harvest is 40% or 400 weight units in Eq. 4.2.

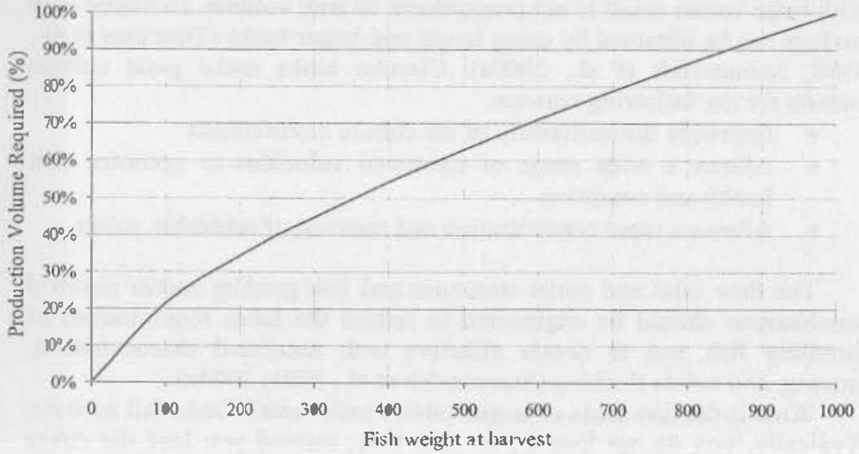


Figure 4.3 Percentage of final tank volume required for the production of fingerlings.

The reader should also be aware at this point that the management methods used to maximize productivity from a given set of tanks can be quite inventive. For example, the simplest management method would be an "all-in and all-out" approach, where a tank is stocked with small fingerlings and then harvested once the fish reach target market size. Clearly, using this approach greatly under utilizes the rearing capacity of the tank, and for that matter, the water conditioning equipment associated with the system to maintain water quality at the highest feeding loads. Watten (1992) presented a mathematical analysis of the effects of sequential rearing on the potential production from the system. Generally, one would probably try to use at least a three phase production strategy from fingerling to market size, i.e. fry production, fingerling production, and growout. Each time fish are moved, though, loss of growth and additional mortality due to stress will occur. It is this loss of productivity that actually limits the number of cohort movements from an economical standpoint, although the time and labor required to perform the movement are also important considerations. Other management methods to maximize the utilization of the RAS system would be to place several tanks on a common water treatment system where the fish are either left in the same tank they were stocked in or the fish are moved from one tank to the next as they increase in size. Any movement of fish is not a good thing and should be avoided by design whenever possible. If unavoidable, every effort should be made to minimize stress during the transfer. This can be accomplished by means of flumes or raceways connecting individual tanks and proper design and layout of the growout facility.

4.3 DESIGN EXAMPLES

Continuing the engineering plan for Omega Industries (OI) proposed construction and operation of an Omega Fish Aquaculture Facility (OFAF), the tanks sizes can be determined based on the anticipated fish biomass stocking densities at harvest or movement from one stage to the next. From Table 3.4, the final fish biomass per tank was 193 kg for the juvenile production, 450 kg for the fingerling and 875 kg for the growout tank. Using Eq. 4.1, the corresponding maximum stocking densities can be calculated, for example the juvenile system:

$$D_{fry} = \frac{L}{C_{density}} = \frac{19.9\ cm}{0.24} = 82.9 \frac{kg}{m^3}$$

Correspondingly, the maximum fingerling tank density is 110 kg/m^3 and 137 kg/m^3 for the growout tank. It is important to remember that these are just recommended maximum densities; the actual density used is a function of operational and management experience and the degree of risk the operator is willing to take, balanced by the economics of the system production. ***It is usually a good idea*** to start out at a lower density, where there is a smaller risk of failure due to operator errors, accidents or misjudgments. Then as operational and management experience is gained, densities can be increased and systems added as needed for oxygen supplementation and monitoring and control. Thus in this design example to limit the need for supplemental oxygen the density in the juvenile tank was set at 30 kg/m^3 (0.25 lbs/gal) in the fingerling tank 40 kg/m^3 (0.33 lbs.gal) and in the growout tank, where supplemental oxygen is required, 50 kg/m^3 (0.42 lbs/gal).

The volume of each tank is equal to the total biomass divided by the stocking density, or as example for the juvenile system:

$$V_{Juvenile} = \frac{Biomass}{Density} = \frac{193\ kg}{30 \frac{kg}{m^3}} = 6.43\ m^3\ or\ 1700\ gal$$

Although in theory tanks of any diameter can be manufactured, fiberglass tanks from major manufactures are normally available in a limited range of sizes. In addition, for ease of management, tank depth is usually less than 1.22 m (4 ft) and to optimize solids removal the diameter to depth ratio should be between 3 and 5. Thus for the juvenile system, a tank with a 3.05 m (10 ft) diameter and a water depth of 0.88 m (2.88 ft), yields a diameter to depth ratio of 4.4 and a volume of 6.4 m^3 (1694 gallons). Table 4.2 summarizes the biomass density and tank dimensions for the three stages used in this example.

Table 4.2 Final Tank Biomass Density and Tank Dimensions for the Three Stage Omega Fish Production Strategy

	Fish Density kg/m^3 (lbs/gal)	Tank Volume m^3 (gal)	Tank Depth m (ft)	Tank Dia. m (ft)
Juvenile:	30 kg/m^3 (0.25 lbs/gal)	6.4 m^3 (1694 gal)	0.88 m (2.88 ft)	3.05 m (10 ft)
Fingerling:	40 kg/m^3 (0.33 lbs/gal)	11.3 m^3 (2978 gal)	1.07 m (3.52 ft)	3.65 m (12 ft)
Growout:	50 kg/m^3 (0.42 lbs/gal)	17.5 m^3 (4621 gal)	1.06 m (3.50 ft)	4.57 m (15 ft)

4.4 Culture Tank Engineering

The production of food fish in large circular tanks has produced large cost savings in comparison to raising the same quantity of fish in more but smaller tanks. Larger circular culture tanks offer many advantages for food fish production. While just a few years ago, an 8 m diameter tank was considered large, now we are seeing 10 and 15 m and even larger diameter tanks being put into production. Substantial savings in both capital and labor costs can be realized by shifting production into fewer but larger culture tanks. Fundamentally, the time it takes to service a small tank or a large tank is similar. In fact, the capital costs associated with large versus small is not proportional to tank volume. Dramatic cost savings can be obtained by using larger and larger tanks (Timmons et al., 1998; Summerfelt et al., 2000a). Circular tanks make good culture vessels for the following reasons:

- Improves the uniformity of the culture environment
- Allows a wide range of rotational velocities to optimize fish health and condition
- Allows a rapid concentration and removal of settleable solids

The flow inlet and outlet structures and fish grading and/or removal mechanisms should be engineered to reduce the labor requirements of handling fish and to obtain effective tank rotational characteristics, mixing, and solids flushing (Summerfelt et al., 2001; 2004a).

What is the downside of larger culture tank units? Tanks fail as units. Typically, you do not lose a part of a tank; instead you lose the entire tank of fish. Distributing a farm's population of fish among multiple tanks provides a measure of protection against entire farm failure. The more fish in a tank, the bigger the economic loss that will occur when a

tank fails. However, as the experience of the management and design team increases, the risk of tank failure decreases, but should never be ignored. It would not be prudent to start out with large culture tank vessels, if this were your first RAS experience. Start small and build upon success. Other challenges of using large circular tanks include:

- Distributing flow to obtain uniform mixing and rapid solids removal
- Grading and harvesting fish
- Removing mortalities
- Isolating the biofilter while treating the fish with a chemotherapeutant

Large tanks are more critically dependent upon tank hydraulic design than are small tanks, because in small tanks, $\leq 1\ m^3$, the overall rate of water exchange tends to be rapid. The rapid hydraulic exchange results in reasonably good water quality, because the high turnover rate (usually as rapid as 10 minutes per tank volume exchange, called hydraulic retention time, HRT) carries more oxygen into the tank and rapidly flushes wastes. Conversely, in large tanks, the HRT tends to be longer (around 45 minutes or greater) and, as a result, the inlet and outlet injection methods and flow rate become dominant factors affecting the uniformity of water conditions in the tank (aside from the feed loading rate). As reviewed in Chapter 3 Mass Balances, the carrying capacity of a tank is influenced by water exchange, feeding rate, oxygen consumption, and waste production. Review and analyze these factors closely before deciding upon your final design.

4.5 Tank Water Velocities

The ability to self-clean is a key advantage of circular tanks. To establish this ability, the water column must be in constant rotation within the tank. The rotational velocity in the culture tank should be as uniform as possible from the tank wall to the center and from the surface to the bottom, and it should be swift enough to make the tank self-cleaning. However, it should not be faster than that required to exercise the fish. Water velocities that are 0.5–2.0 fish body lengths per second are optimal for maintaining fish health, muscle tone, and respiration (Losordo and Westers, 1994). Velocities required to drive settleable solids to the tank's center drain should be greater than 15 to 30 cm/s (Burrows and Chenoweth, 1970; Mäkinen et al. 1988). For tilapia, Balarin and Haller (1982) reported an upper current speed of 20–30 cm/s.

For salmonids, Timmons and Youngs (1991) provided the following equation to predict safe non-fatiguing water velocities:

$$V_{safe} < \frac{5.25}{L^{0.37}} \tag{4.3}$$

In circular tanks, velocities are reduced somewhat away from the walls, which allow fish to select a more comfortable water velocity, as compared to raceway designs where velocities are uniform along the channel.

A phenomenon that has been observed by all culturists familiar with raceways is the fact that, more often than not, fish concentrate themselves in the upper one-third of the raceway towards the inlet, while sparsely occupying the lower two-thirds. This is, of course, a function of fish density, but unless densities are ~80 kg/m^3 or greater, then raceway space is often poorly utilized. Higher densities require higher exchange rates or the injection of pure oxygen. Standard length raceways of 20 m to 40 m should be operated at six to four exchanges per hour respectively, but often are not. Even at these relatively high water exchange rates, R, the velocities are still below 5.0 cm/sec as Eq. 4.4 shows:

$$V_{raceway} = \frac{L_{raceway} \cdot R}{36} \tag{4.4}$$

The constant in Eq. 4.4 is the number of seconds per hour divided by 100 to convert meter (length or distance the water must travel) to cm, the unit used to express the velocity. Such velocities are well short of cleaning velocities of 15 to 30 cm/sec or recommended velocities for fish conditioning, which range from 0.5 to over 2.0 times their body length per second (Poston et al. 1969; Woodward and Smith, 1985; Needham, 1988; Josse et al. 1989; Timmons and Youngs, 1991).

Timmons and Youngs (1991) pointed out such deficiencies and concluded that, in practice, raceways are managed much closer to their design requirement for oxygen supply than for cleaning requirements. The means to overcome this deficiency is to either design very long raceways, or raceways having a very small cross-sectional area, neither of which is very practical. To at least partly overcome this drawback with standard length raceways, Boersen and Westers (1986) propose the use of baffles. These are evenly spaced throughout the raceway at distances approximately equal to the width of the raceway. The gap or space between the lower edge of the baffle and the bottom of the raceway dictates the velocity relative to the exchange rate. Their main

function is to make the tank self-cleaning, removing solid waste as soon as it settles, thus preventing buildup and subsequent resuspension through fish activity. The relatively intact solids will settle quickly behind the fish retaining screen in a small section of the raceway dedicated to that function. This simple technique provides for an effective solids management approach. However, baffles increase velocities only over a small cross-sectional area of the pond. Fish will utilize this relatively high velocity zone but since the space is so limited, there is room only for a small percentage of the population. Yet it is desirable, especially for salmonids, to expose all fish to relatively high water velocities.

Using Eq. 4.3 for a 10.0 cm fish, the V_{safe} velocity should not exceed 2.2 times its body length per second (BL/sec); for a 20.0 cm fish it is 1.73 BL/sec. Totland et al. (1987) exercised large Atlantic salmon (56.3 cm and 2,038 g) at velocities of 0.40 to 0.45 BL/sec. They found improved survival rates over caged fish except during the initial two week adjustment period when losses were 1.2%, much greater than the reference group. Final losses were 4.4% versus 8.8%, and the weight gain was nearly 40% greater in the exercised fish. According to industry standards, quality was rated 9.2% higher. Based on Eq. 4.3, the recommended velocity for this size fish would be 1.2 BL/sec but favorable results were obtained at the lower velocities of 0.45 BL/sec, which represents a velocity of 25.3 cm/sec, far above raceway velocities.

Needham (1988) recommends velocities between 0.5 and 1.0 BL/sec for Atlantic salmon. Woodward and Smith (1985) exercised rainbow trout at velocities of 1.5 BL/sec for 42 days. It improved fish quality in terms of better stress resistance. Indeed, it has been shown that sustained swimming speed improves disease resistance. Leon (1986) found this to be true along with improved growth rates and feed conversions when brook trout were reared in velocities of 1.5 to 2.0 BL/sec.

Josse et al. (1989) maintained rainbow trout at sustained velocities of 2.5 BL/sec with bursts of 3.8 BL/sec for a few minutes daily. This latter velocity was aimed at developing the white musculature. The continuous cruising speed served to develop the red musculature, but it was found to also stimulate the white musculature without exhausting the fish. Continuous swimming speed also had a positive effect on tail musculature development. The red musculature increased by 27% and the white by 9% over the control groups maintained in still water. The authors also concluded that the permanent rotary water movement (the direction of the water needed to be reversed from time to time to prevent uneven muscle development) ensured a perfect homogenization of the medium, encouraged optimal distribution of the fish, and inhibited their

territorial behavior which, in turn, resulted in a 100% increase in rearing density over the control group (68.4 versus 36.0 kg/m^3). Mortalities during the early rearing phase were significantly less in the high velocity environment, and the experimental fish grew as well as the control group, despite the sustained swimming action. Much earlier studies by Poston et al. (1969) pointed out such benefits. Brook trout, exposed to velocities in excess of 2.0 BL/sec, showed increased stamina and a faster rate of replacement of muscle glycogen after exposure to strenuous exercise in a stamina tunnel, compared to unconditioned fish. The conditioned fish also showed a more efficient rate of food conversion.

Forced exercise, contrary to what one would expect, seems to result in reduced oxygen consumption. This has been attributed to physiological adaptations, such as increased white muscle activity, improvements in cardiac output, and enhanced oxygen carrying capability of the blood (Woodward and Smith, 1985). There can also be savings in "breathing" cost. Fish who can maintain their position in fast-flowing water need only to open their mouths to ventilate their gills. This is termed ram ventilation. Ram ventilation can contribute to saving energy in two ways. The fish does not have to pump water over its gills which, in turn, result in less turbulence, i.e., a more streamlined flow of water is maintained over the body that reduces the need to make continual positional adjustments. This hydrodynamic advantage results in small, but measurable, reductions in oxygen consumption.

The cost of "breathing" in a dense medium, such as water, can be substantial. The costs, in terms of total oxygen uptake, can be from 10% (Jones and Randall, 1978) to 20% or more (Shelton, 1970) and, according to Schumann and Piper (1966) as much as 30%. Watten and Johnson (1990) offer yet another explanation. The elevated surface velocities in their cross-flow rearing tank, along with a homogeneous DO concentration, may accelerate the diffusion of oxygen at the air-water interface, thus making more oxygen available than the net decrease shows.

Round tanks, in contrast to raceways, do not have a distinct water quality gradient and frequently the rearing environment is considered to be homogeneous. Colt and Watten (1988) describe the ideal round tank as a continuous-flow stirred tank reactor, where the dissolved gas concentrations are well mixed and equal to the concentration in the effluent. However, Tvinnereim and Skybakmoen (1989) point out that in a complete mixed flow reactor, the maximum possible water exchange will be 63.2% during the theoretical mean retention time. Nevertheless, high concentrations of dissolved oxygen (DO) entering a round tank are rapidly diluted with lower DO water in the rearing unit, which is very

different from raceways. Since the round tank has no gradient, ammonia and carbon dioxide are fully mixed into the rearing environment, resulting in the continuous presence of some level, in contrast with raceways. One must make sure that such continuous exposure to these potentially detrimental conditions is countered with healthy dissolved oxygen concentrations.

Water velocities in round tanks are, to a large degree, controllable independently of the water exchange rate (Davidson and Summerfelt, 2004), in contrast with raceways. The most critical factors are the design of the inlet and outlet arrangements, the flow through the tank, and the flow through the center drain (Tvinnereim and Skybakmoen, 1989; Summerfelt et al., 2000b; Davidson and Summerfelt, 2004). Properly designed inlet intake-outlet provisions also contribute to self-cleaning characteristics. Circular tanks have a number of important advantages over raceways. Round tanks provide cheaper rearing space and they can be operated at low water exchange rates while still creating desirable velocities, hydraulic patterns and self-cleaning characteristics. Round tanks can also receive very high DO inputs without creating "hot spots" and they can be easily equipped with feeders, requiring only one or at most a few feeding stations. With round tanks it is often possible to accomplish the maximum cumulative oxygen consumption (COC) within a single rearing unit. In other words, the water is "used up" after passing through one tank instead of a series of two or three. All units can, therefore, be placed on the same elevation level.

4.6 Round Tanks

Tanks are designed with considerations given to production cost, space utilization, water quality maintenance, and fish management. Tank designs used for intensive fish culture have been presented frequently and reviewed in the literature (Wheaton, 1977; Klapsis and Burley, 1984; Piper et al. 1982; Cripps and Poxton, 1992; Timmons et al., 1998; Summerfelt et al., 2000a). There is a definite trend towards large circular culture tanks for food fish production. Tanks larger than 10 m in diameter, which used to be referred to as pools, are now reasonable choices for culture systems in intensive indoor operations.

Circular tanks are attractive for the following reasons (Timmons et al., 1998):

- Simple to maintain
- Provide uniform water quality

- Allow operating over a wide range of rotational velocities to optimize fish health/condition
- Settleable solids can be rapidly flushed through the center drain
- Designs that allow for visual or automatic observation of waste feed to enable satiation feeding are possible

Recommended tank diameter to depth ratios vary from 5:1 to 10:1 (Burrows and Chenoweth, 1955; Chenoweth et al. 1973; Larmoyeux et al. 1973); even so, many farms use tanks with diameter:depth ratios as low as 3:1. The authors favor tanks with diameter depth ratios that are less than 5:1. Selection of a tank diameter:depth ratio is also influenced by factors such as the cost of floor space, water head, fish stocking density, fish species, and fish feeding levels and methods (Timmons et al., 1998). Choices of depth should also consider ease of workers handling fish within the tank and safety issues of working in waters that may be more than "chest" high.

In the early years of RAS, tanks that were actually deeper than their diameter were touted as a key design factor to economic success. None of these 'silo' systems were successful, mostly for problems related to fish management. Even in more modest attempts to utilize deeper tanks, e.g., a 3:1 ratio of diameter:depth, not all fish will effectively distribute into the entire water column. An example of this species specific attribute includes tilapia, which utilize the entire water column, versus walleye or flounder, which are surface area animals and do not utilize the water column nearly as effectively as tilapia.

Inlet Flow Structures

Circular tanks can achieve relatively complete mixing (Davidson and Summerfelt, 2004), i.e., the concentration of a dissolved constituent in the water flowing into the tank changes instantaneously to the concentration that exists throughout the tank. Therefore, if adequate mixing can be achieved, all fish within the tank are exposed to the same water quality. Good water quality can be maintained throughout the circular culture tank by optimizing the design of the water inlet structure and by selecting a water exchange rate so that the limiting water quality parameter does not decrease production when the system reaches carrying capacity (Timmons et al., 1998).

The water inlet and outlet structures and fish grading and/or removal mechanisms should be engineered to reduce the labor requirement for fish handling and to obtain uniform water quality, rotational velocities, and solids removal within the circular tank. There is *nothing more*

irritating than pipes and other devices that are in the way, when it is time to harvest a tank. Such obstructions also may decrease the effectiveness of the self-cleaning attributes of round tank culture in the first place. It is useful to visually inspect a tank being operated without fish to ensure that the tank water is effectively rotating and the presence of any dead zones can be noted and eliminated. Also, inlet structures can be installed for ease of removal during harvesting procedures, when generally supplemental aeration devices are operated as well.

"Rule of Thumb"

There is *nothing more irritating* than pipes and other devices that are in the way when it is time to harvest a tank.

Circular tanks are operated by injecting water flow tangentially to the tank wall at the tank outer radius so that the water spins around the tank center, creating a primary rotating flow. The no-slip condition that exists between the primary flow and the tank's bottom and side walls creates a secondary flow that has an appreciable inward radial flow component at the tank bottom and an outward radial flow at the tank surface, Fig. 4.4. This inward radial flow along the bottom of the tank carries settleable solids to the center drain and thus creates the self-cleaning property so desired in circular tanks. Unfortunately, in a circular tank with such flow, an area near the center drain region will become an irrotational zone with lower velocities and poor mixing, resulting in solids settling onto the floor. The magnitude of the irrotational zone depends on the introduction of tangential flows near the walls, the diameter:depth ratio, and the overall rate of flow leaving the center bottom drain (Tvinnereim and Skybakmoen, 1989; Timmons et al., 1998; Summerfelt et al., 2000a; Davidson and Summerfelt, 2004). Because this irrotational zone has lower water velocities and does not mix well, it can decrease the effective use of the culture tank by producing short circuiting of flow, by creating localized water quality gradients (especially of concern are reduced oxygen levels), and by providing a quiescent zone where solids can settle and collect.

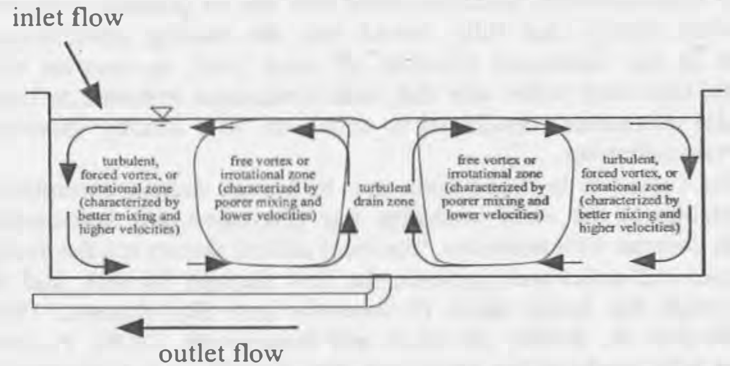


Figure 4.4 The primary rotating flow (not shown, but created by injecting the flow tangential to the tank wall) creates a secondary rotation that flows radially (shown here) and carries settleable solids towards the tank's bottom center drain in a phenomenon called the tea-cup effect (from Timmons et al., 1998).

The degree to which a circular tank can self-clean is in part related to the overall rate of flow leaving the center drain (Summerfelt et al., 2000b; Davidson and Summerfelt, 2004). Further, effective solids removal also depends upon the fish re-suspending the settled materials through their movements (Summerfelt et al., 2000b). This explains in part why tanks with low fish biomass do not clean as well as tanks with higher biomass. In addition, because aquaculture solids have specific gravities that are relatively close to that of water (typically 1.05–1.20; Chen et al. 1993; Potter, 1997), sloping the floor towards the center drain does not improve the self-cleaning attributes of a circular tank. Sloped floors are useful only when a circular tank is drained for maintenance purposes.

Rotational velocity can be controlled by design of the water inlet structures, so water flow does not have to be increased beyond that required for the fish's culture environment (Klapsis and Burley, 1984). Tvinnereim and Skybakmoen (1989) reported that the current velocity in a tank can be largely controlled by varying the inlet impulse force (F_i), which is defined as:

$$F_i = \rho \cdot Q \cdot (V_{orif} - V_{rota}) \tag{4.5}$$

The inlet impulse energy largely dissipates as it creates turbulence and rotation in the rotational zone, Fig. 4.4. The impulse force, and thus the rotational velocity in the tank, can be regulated by adjusting the inlet

flow rate or the size and/or number of inlet openings. Paul et al. (1991) reported that the tank rotational velocity (assumed to mean at the outside edge) is roughly proportional to the velocity through the openings in the water inlet structure:

$$V_{rota} \approx \alpha \cdot V_{orif} \tag{4.6}$$

where the proportionality constant (α) is generally from 0.15-0.20, depending on the design of the inlet flow structure; others report an α of 4 to 6% for overall average velocity of tank flow.

The manner of flow injection has been shown to influence (Skybakmoen, 1989; Tvinnereim and Skybakmoen, 1989; Davidson and Summerfelt, 2004):

- Uniformity of the velocity profile through the tank;
- Strength of the secondary radial flow along the tank bottom towards the center drain, i.e., the ability of the tank to move settleable solids to the center drain; and
- Uniformity of water mixing.

Using Fig. 4.5, you can predict the velocity needed from the inlet orifices to create the desired tank water velocities. By installing a site tube immediately above the first orifice hole in the vertical inlet pipe, you can observe the available head to create velocity. Assuming the tank water velocities near the outside walls will be equal to 15 or 20% of the orifice velocity as noted above, you can adjust the number of holes and hole area in the inlet pipe to create the necessary velocities to achieve the desired tank water velocities. Note that in 'Cornell-type' dual-drain tanks, the average tank velocity will be substantially less than the velocities near the outer walls and thus obtaining satisfactory velocities as you move towards a center drain becomes a function of the flow discharged through the center of the tank (Davidson and Summerfelt, 2004). You can estimate the water rotational velocity about the perimeter of the tank "on-paper" before you construct the inlet pipes using a method described by Labatut et al. (2007). If the site tube is located immediately above the first orifice hole, the required dynamic heads, H, to create the velocity desired is 1.5 H ($H = V^2/2g$; V is velocity at the orifice and g is the acceleration due to gravity, 9.8 m^2/s; see Chapter 12 for more information on fluid mechanics).

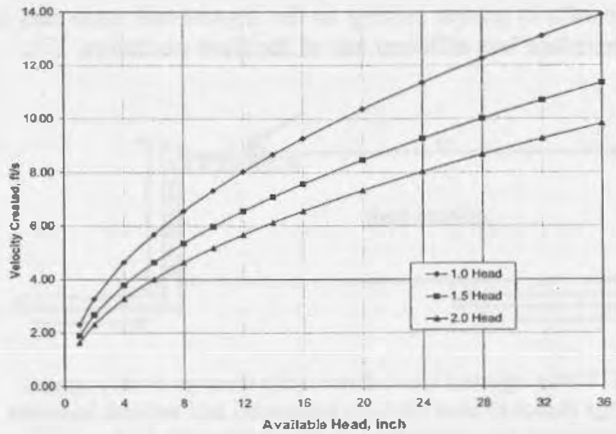


Figure 4.5 Resulting orifice velocities for a specific head available. Typical inlet orifice will require 1.5 dynamic heads (from Labatut et al., 2007).

Skybakmoen (1989) and Tvinnereim and Skybakmoen (1989) compared the tank hydraulics that resulted from injecting the water flow tangentially at the outer radius of the circular tank with either:

- a traditional open-ended pipe point source,
- a short, horizontal, submerged, distribution pipe with its axis oriented towards the tank center and with evenly spaced openings along its length (directed at 30° below the water surface),
- a vertical submerged distribution pipe with evenly spaced openings along its length, and
- and an inlet flow distribution pipe that combines both vertical and horizontal branches.

Open-ended pipe inlets create non-uniform velocity profiles in the tank, e.g., much higher velocity profiles along the tank wall; poor mixing in the irrotational zone, resuspension of solids throughout all tank depths; and poor flushing of solids from the bottom (Skybakmoen, 1989; Tvinnereim and Skybakmoen, 1989). Horizontal submerged pipe inlets improve water mixing effectively throughout the tank, but create a weaker and less stable bottom current (for solids cleaning). Vertical submerged inlet distribution pipes provide better self-cleaning than when injecting the water flow through an open-ended pipe or a horizontal distribution pipe, but the stronger bottom current (responsible for particle

removal) also results in poorer mixing in the irrotational zone and short circuiting and therefore less efficient use of the flow exchange.

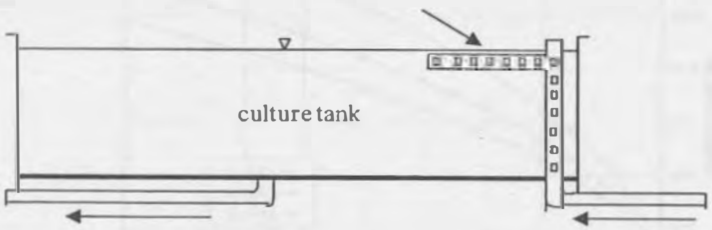


Figure 4.6 Water injected into culture tanks through evenly spaced openings (holes or slots) in both horizontal and vertical injection pipes produces more uniform rotational velocities both radially and vertically, more uniform mixing, and better solids flushing (from Timmons et al., 1998).

Maximum uniformity in tank water conditions can be obtained by using an inlet flow distribution pipe that combines both vertical and horizontal branches, Fig. 4.6. The inlet pipes must be placed somewhat away from the wall so that fish can swim between the pipe and wall. This design approach is an effective way to:

- Achieve uniform mixing,
- Prevent short circuiting of flow,
- Produce uniform velocities along both the tank's depth and radius, and
- Effectively transport waste solids to the tank bottom and out the center drain.

For large circular tanks, e.g., diameter > 6 m, placing multiple flow distribution pipes at different tank locations can improve solids removal, velocity uniformity, and water quality homogeneity. Again, perform visual observations prior to fish placement. Recent experience indicates that by using only vertical inlet pipes and then pointing the exhaust jets at approximately 45° from the tank wall is reasonably effective. This may produce a flow pattern in the tank effective enough to eliminate the need for any horizontal inlet pipes. The horizontal pipes will interfere more with harvesting and sampling activities than would just the vertical pipes near the tank wall. More elegant solutions to inlet distribution have been presented in the literature, e.g., Watten and Johnson, 1990; Davidson and Summerfelt, 2004. Flow inlet structures can be built

directly in the wall of fiberglass culture tanks, which eliminates the need for vertical or horizontal pipe inlet structure and their inlet nozzle orientation and direction can be readily adjusted to provide an effective method for controlling water rotational velocity (Davidson and Summerfelt, 2004).

The reader is referred to the extensive literature on circular tank design and inlet structures for further information: Burrows and Chenoweth, 1955; Larmoyeux et al., 1973; Wheaton, 1977; Klapsis and Burley, 1984 and 1985; Skybakmoen, 1989; Tvinnereim and Skybakmoen, 1989; Paul et al., 1991; Goldsmith and Wang, 1993; Timmons et al., 1998; Summerfelt et al., 2000a; 2000b; 2001; Davidson and Summerfelt, 2004.

Outlet Flow Structures

Circular fish culture tanks concentrate settleable solids, e.g., fecal matter, feed fines, and uneaten feed, at their bottom and center. The tank center is then the logical location for the bottom drain. The bottom center drain should be designed to continuously remove the concentrated settleable solids and for the intermittent removal of dead fish that are captured at the bottom center drain. The bottom center drain structure is also used for water level control by connecting it to a weir, either on the inside, Fig. 4.7, or the outside of the tank, Fig. 4.8.

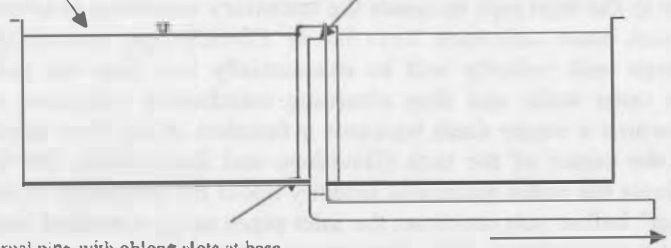


Figure 4.7 Internal standpipe arrangement. An internal standpipe made of two pipes can be used to both control depth and remove solids from the tanks bottom: the outer pipe is used to pull flow from the bottom of the tank and the inner pipe is used to set the water depth within the tank.

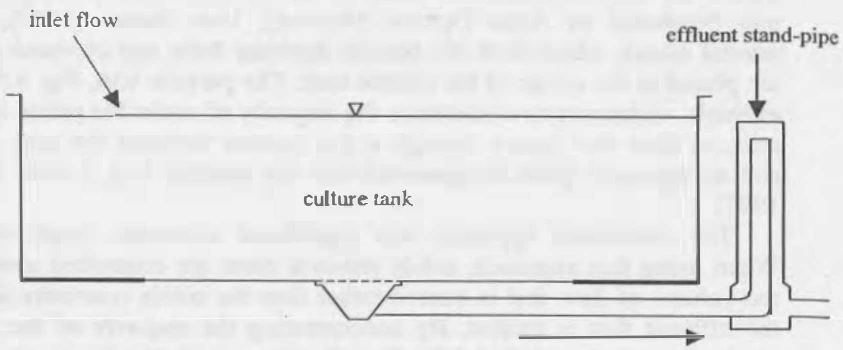


Figure 4.8 External standpipe arrangement. Excluding fish from the center drain with a bottom perforated plate or screen requires use of an external standpipe to control water depth, but makes the standpipe easily accessible and avoids obstructions in the center of the tank.

When a center standpipe is used on the inside of a tank, it can be designed so that it either captures and stores settleable solids near the drain (where they can be flushed at intervals) or it continuously withdraws settleable solids from the bottom of the culture tank. To continuously withdraw settleable solids through a center standpipe that also controls water depth requires use of two concentric pipes. Perforated slots at the base of the outer pipe (Larmoyeax et al., 1973) or a gap at the base of the outer pipe (Surber, 1933) forces flow to be pulled from the bottom of the tank (capturing settleable solids) and the inner pipe is used as a weir to set the water depth within the tank, Fig. 4.7.

As early as 1933, the use of self-cleaning center standpipe had been presented in the literature (Surber, 1933 & 1936). Figure 4.7 shows the recommended design for creating an adjustable gap between the bottom of the outer pipe and the tank bottom in order to increase suction while forcing the flow to leave at the tank bottom where settleable solids collect. The distance between the two pipes, i.e., the annular space should be selected to create a velocity large enough (0.3 to 1.0 m/s depending upon the size and density of the particles) to entrain solids up to the top of the inner pipe. A patented method that incorporates these principles uses an annular approach plate to enhance particle entrapment, Fig. 4.9.

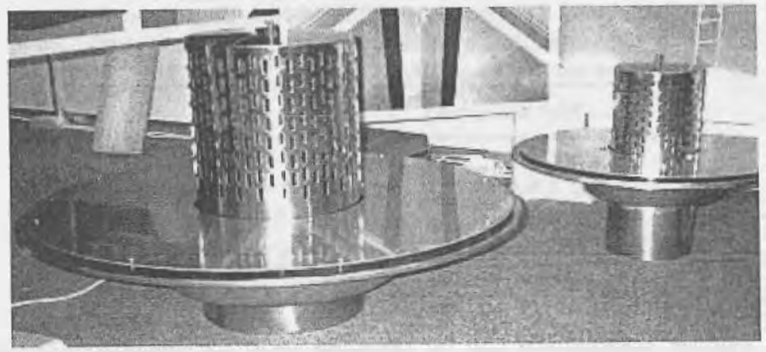


Figure 4.9 A solid annular approach plate fixed above the bottom center drain as developed and patented (U.S. Patent No. 5,636,595) by Lunde et al. (1997) to exclude fish and enhance particle removal by accelerating particles into the drain (from Summerfelt et al., 2001).

When an external standpipe controls water depth, a perforated plate or screen, Fig. 4.8, can be used to cover the bottom center drain; this allows solids to leave the tank but excludes fish. Corrosion-resistant screening material, such as perforated sheets of aluminum, stainless steel, fiberglass, or plastic is used to cover drain outlets. We recommend using perforated screening with horizontal oblong slots instead of holes, because the slots are easier to clean, provide greater open area, and do not clog as readily as round holes (Piper et al., 1982; Timmons et al., 1998). Guidelines for slot size based upon fish size are given in Table 4.3. Ideally, openings through the screen covering the center drain should be small enough to exclude fish and yet large enough not to become clogged with feed pellets or fecal matter. Entrapment of fish on the outlet occurs when fish cannot escape the area in front of the drain because the water velocity in that area is too great. Fish impingement is minimized by providing a total open area through the outlet screen so that the water velocity through the screen is ≤ 30 cm/s. Depending upon the species and life stage, certain situations particularly with smaller fish require water velocities ≤ 15 cm/s (Pankratz, 1995), e.g., see Eq. 4.3. These velocities do not produce a significant pressure drop through the screen openings, thus minimizing fish impingement.

Table 4.3 The Horizontal Oblong Slot Size Depends Upon the Size of Fish to be Retained (Fish Species was not Specified, from Piper et al. 1982)

Slot Size (mm)	Fish size, g
1.6 x 3.2	fry to 0.45 g
3.2 x 6.4	0.45 to 2.3 g
6.4 x 12.7	2.3 to 15 g
12.7 x 19.1	15 g and larger

Further information can be obtained from the literature on outlet designs: Surber, 1933 and 1936; Burrows and Chenoweth, 1955; Larmoyeux et al. 1973; Piper et al. 1982; Klapsis and Burley, 1985; Josse et al. 1989; Skybakmoen, 1989; Tvinnereim and Skybakmoen, 1989; Pankratz, 1995.

4.7 Cornell Dual-Drain Design

Circular fish culture tanks can be managed as swirl settlers, i.e., settling basins with two effluents, because of their capability to concentrate solids at their bottom and center. Solids that concentrate at the bottom center can be removed in a small flow stream by using a bottom-drawing center drain, while the majority of flow is withdrawn at an elevated drain. Cobb and Titcomb (1930) and Surber (1936) were the first to report the use of a second bottom-drawing drain to remove solids that were settled in the center of circular culture tanks. More recently, settled solids have been reportedly concentrated in 5–20% of the total flow that leaves the bottom center drain of circular culture tanks when the remainder of the flow leaving the tank (roughly 80–95% of the total) was withdrawn through a fish-excluding port located above the bottom-drawing drain (Mäkinen et al. 1988; Eikebrokk and Ulgenes, 1993; Lunde et al. 1997; Timmons et al., 1998; Summerfelt et al., 2000b; Davidson & Summerfelt, 2004). There have been at least two patents filed on double-drain designs:

- Lunde et al. (1997), *Particle trap*, US Patent # 5636595.
- Van Toever (1997), *Water treatment system* US Patent # 5593574.

The location of the two tank drains have typically been at the center of the tank, which then takes advantage of both the 'tea-cup effect' and the strength of the overall flow when it drains through the tank center.

This is the approach has been taken by the manufacturers of the particle trap (marketed by Aqua Optima, Norway), Van Toever (1997), and several others, where both the bottom drawing drain and elevated drain are placed at the center of the culture tank. The particle trap, Fig. 4.9, for example, endeavors to concentrate the majority of settleable solids into a reduced flow that passes through a gap created between the tank floor and an approach plate incorporated into the particle trap (Lunde et al. 1997).

The dual-drain approach has significant economic implications. When using this approach, solids removal costs are controlled more by the volume of flow that is treated rather than the solids concentration of the effluent that is treated. By concentrating the majority of the solid wastes in only 5 to 20% of the flow leaving a tank via the center low-flow drain, treatment costs are proportionately reduced while treatment efficiency is increased (Summerfelt et al., 2001; Davidson and Summerfelt, 2005).

The Cornell-type dual-drain culture tank, Figs. 4.10 and 4.11, differs significantly from the other dual-drain designs, because it is the only design that removes the majority of flow through an elevated drain located on the tank's sidewall; all other double-drain approaches place both drains in the tank center.

Using the Cornell-dual drain design basically reduces the intensity of the center vortex, which can be exceptionally strong when all of the flow is extracted from the center of round tanks, especially when tank exchange rates approach or exceed one exchange per hour. In these situations, the center vortex can be strong enough to generate an upward water flow velocity at the tank center with sufficient force to pull solids back up into the water column, causing settled solids to "plume" up (Davidson & Summerfelt, 2004). The Cornell-dual drain also reduces the velocities near the center drain because the flow leaving the center drain is only 5 to 20% of the total flow from the tank (Davidson & Summerfelt, 2004). As a result, fish distribution is much more uniform in a Cornell dual-drain compared to conventional double drains or single drain tanks that are operated at tank exchange rates of ≥ 1 exchange per hour. This is a key advantage because the fish effectively utilizes the entire tank volume. In a conventional dual-drain tank, it is not uncommon to see the center third of the tank being unoccupied by fish.

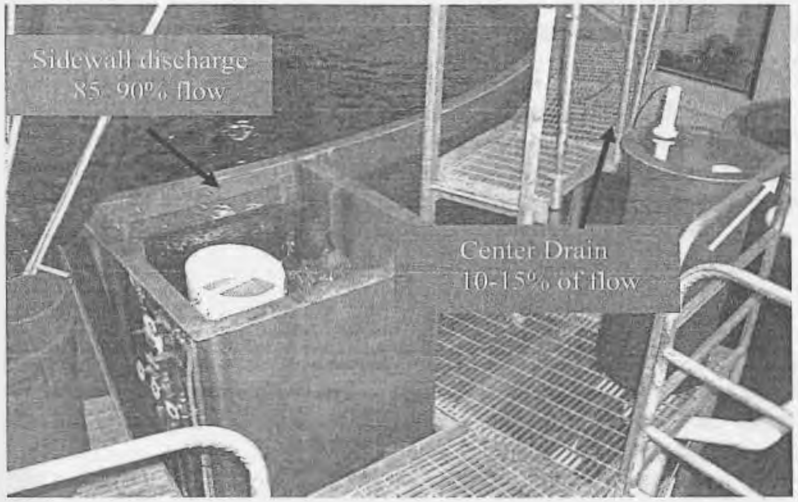


Figure 4.10 Cornell Dual Drain system (40,000 gallon tank, total flow of 1,200 gpm, from Freshwater Institute).

The Freshwater Institute (Shepherdstown, WV) and Cornell University recently completed a study of the Cornell dual-drain design (Summerfelt et al., 2000b). The Freshwater Institute used rainbow trout at densities of 60 kg/m^3 and fish were fed 1% body weight per day; Cornell used 500 g tilapia on full feed at a tank density of 90 kg/m^3. Settleable solids fractionation and tank mixing were studied using a 3.66 m (12 ft) i.d. x 1.22 m (4 ft) tall circular Cornell-type dual-drain tank at the Freshwater Institute (FI) and a 2 m i.d. x 1 m (3.2 feet) tall (6.5 ft) tank at the Cornell site. A range of hydraulic exchange rates (1 to 7 tank volumes per hour) and percentage bottom drain flow (5, 10, and 20% of total flow) were used through the tank.

"Rule of Thumb"

Choose the Center Drain Flow as the largest of:
a) 6 Lpm/m^2 (0.15 gpm/ft^2) of floor
b) HRT center drain < 200 minutes, or
c) 10 to 15% of total tank flow rate.

The solids removal capabilities of the Cornell-type dual-drain design have been determined in actual tests with and without fish present and at a variety of tank sizes. Sinking PVC cylindrical beads (3 mm in diameter, with a specific gravity of 1.05) were added to the culture tank

with fish present. Replicated bead flushing tests were used to determine the influence of hydraulic exchange rate (1 and 2 rearing tank volume exchanges per hour), diameter-to-depth ratio (12:1, 6:1, 3:1), and the percentage of flow leaving the bottom center drain (5, 10, and 20% of total flow) on solids flushing characteristics (Summerfelt et al., 2000b). The bead flushing data was analyzed using the solution to a non-steady-state mass balance and used to quantify the amount of solids flushing that occurs through the bottom center drain in comparison to that which was due to simple dilution versus solids transport produced by settling and radial flow dynamics (Summerfelt et al., 2000b).

Total suspended solids (TSS) concentrations were also measured in bottom and sidewall discharges and in the effluent from a commercial microscreen filter treating the bottom flow. Mean results from data collected at the Freshwater Institute indicated that the Cornell-type double-drain tank concentrated the majority of TSS in the bottom flow. The TSS concentration discharged through the bottom drain averaged 19.6 ± 3.6 mg/L (± standard error), while the TSS concentration discharged from the sidewall averaged only 1.5 ± 0.2 mg/L (Summerfelt et al., 2000b). This represents a 13 fold increase in solids concentration from the center drain. Practically speaking, the side wall drain discharges the best water quality in terms of suspended solids, since the solids are constantly moving towards the center drain and influent water also enters from the outside edge of a tank. The sidewall discharge, which represented 80–95% of the total flow, but only 1.5 mg/L TSS, would probably not require any further treatment and most likely could be discharged directly under most state and federal regulations. Further treatment of the bottom discharge across a commercial microscreen filter then captured 82% (± 4%) of the TSS in the bottom discharge, so that only an average of 3.5 ± 0.8 mg/L TSS would be discharged with the bottom flow (Summerfelt et al., 2000b). In a recirculating system, Davidson and Summerfelt (2005) found that radial flow settling units could be used to remove approximately 80% of the TSS from the water exiting the bottom-center drain of large 'Cornell-type' dual-drain tanks.

Further research was conducted by Davidson and Summerfelt (2004) to determine how bottom drain flow rate affects solids flushing, water mixing, and water velocity profiles within large, i.e., 10 and 150 m^3 circular 'Cornell-type' dual-drain culture tanks. Results showed relatively uniform water mixing was achieved in both the 10 m^3 and the 150 m^3 'Cornell-type' dual-drain culture tanks when tested at high fish densities (90 to 98 kg/m^3) and at hydraulic exchange rate of one tank volume every 20-32 minutes. Mixing efficiency was largely attributed to the high density of fish and was not dependent on the percent of tank

flow discharged through the bottom-center drain flow. The rate that settleable solids flushed from each culture tank was found to be dependent on the water flow exiting the bottom-center drain and on the rotational period of the water within the tank. Optimal water velocities for rapidly flushing solids from the 'Cornell-type' dual-drain tanks were found to be a rotational period of 1.3 to 1.7 minutes and a water flow through the tank's bottom-center drain of at least 5-6 Lpm for every 1 m^2 of tank plan area, i.e., 0.12-0.15 gpm/ft^2 at 2 tank volume exchanges per hour (Davidson and Summerfelt, 2004). Later research at the Freshwater Institute indicated that higher surface loading rates are required to rapidly flush feed when the tank exchange is reduced to 1 exchange per hour (larger HRT's), hence the need to use all three criteria given earlier in the above "Rule of Thumb" box, and using the criterion that resulted in the largest flow rate.

When using the Cornell Double-Drain approach, the bottom flow is only 5–20% of the total tank discharge, so it would be possible and practical to further treat this flow with finer microscreen filters or by using settling tanks, wetlands, or sand filters, if required to meet stringent state or federal discharge regulations. Therefore, conventional flow-through systems that use circular culture tanks installed with the Cornell-type dual drain configuration can capture more of the waste solids and phosphorus from the water flow than can be captured within typical raceways operation that contain quiescent zones and off-line settling ponds. Additionally, when the Cornell-type dual drain tank is used within partial-recirculating systems, these systems are capable of supporting high production densities while providing uniformly healthy water quality, optimized water velocities, and efficient, rapid, and gentle solids removal (Summerfelt et al., 2004b). This type of partial recirculating system would be able to capture approximately 80% of the total waste solids produced (Summerfelt et al., 2004b), which is significantly better waste capture than is typically achieved within raceways operated under serial water reuse, e.g., 50% solids capture (Mudrak, 1981). Also, the solids filter treating the discharge from the partial-reuse system could be relatively small and inexpensive in comparison to a similar unit scaled to treat the much larger flow discharged from a raceway operation of relatively similar annual fish production (Summerfelt et al., 2004b). These combinations of treatment methodology offer a powerful approach to effective and efficient waste stream control from intensive culture systems.

THEORETICAL BASIS FOR SOLIDS REMOVAL AND PRACTICAL APPLICATION

The ideal theoretical mean residence time (MRT_{ideal}), meaning with perfect and uniform mixing, or hydraulic retention time (HRT) can be calculated using the water volume in the tank (V_{tank}) and the water flowrate through the tank (Q):

$$MRT_{ideal} = \frac{V_{tank}}{Q} \tag{4.7}$$

Solids will be removed from a tank based upon mass action or simply HRT and an enrichment factor (k) due to concentration of solids near the center drain (Summerfelt et al., 2000b). The relative importance of these two solids flushing mechanisms can be described as the enrichment factor as a percent (φ_{enrich}):

$$\varphi_{enrich} = \frac{k}{k + \dfrac{Q}{V_{tank}}} \cdot 100 \tag{4.8}$$

Solids removal in the absence of incoming, generation, or accumulation of solids can be described in an unsteady-state mass balance broken into a component representing simple mass action of flow to the bottom drain and a component representing solids enrichment at the bottom drain due to a combination of sedimentation and radial flow, assuming that the solids outflow at the tank's sidewall is negligible (Summerfelt et al., 2000b). This is done to distinguish between the two different mechanisms transporting solids to the bottom center drain:

Loss = – Outflow (mass action) – Outflow (enrichment) (4.9)

Or more specifically,

$$V_{tank} \cdot \frac{dC}{dt} = -Q_{out,b} \cdot C_{out,b} - k \cdot C_{out,b} \cdot V_{tank}$$
$$= V_{tank} \cdot \left\{ -\left(\frac{Q}{V_{tank}} + k \right) \cdot C_{out,b} \right\} \tag{4.10}$$

Integration provides an equation that can be used to model solids removal through the bottom drain in real time:

$$C_{out,b}(t) = C_{out,b}(t=0) \cdot exp\left[-\left(\frac{Q}{V_{tank}} + k\right) \cdot t\right] \qquad (4.11)$$

Note that the flushing of a homogeneously distributed pollutant is only due to mass action (Summerfelt et al., 2000b), i.e., the culture tank exchange rate Q/V:

$$C(t) = C(t=0) exp\left[-\left(\frac{Q}{V_{tank}}\right)t\right] \qquad (4.12)$$

Now, it can be seen in the above equations how the k-value (1st order enrichment constant) increases the rate of solids flushing relative to the culture tank exchange rate.

The k-values estimated from the pellet-tracer tests are an indication of the rate that solids are flushed from the bottom-center drain and also of the relative strength of the radial flow. Strong radial flows and rapid solids flushing were created using 2 ex/hr and diameter:depth ratios of 3.1:1 and 6:1 in the Freshwater/Cornell trials (Summerfelt et al., 2000b). However, a very important observation was that solids removal was not effective at low exchange rates. At one ex/hr, the settleable solids frequently deposited near but did not exit the tank's bottom-center drain. As the diameter:depth ratios became larger (more shallow tanks), the removal rates became less effective (Summerfelt et al., 2000b). A tank with a diameter:depth ratio of 12:1 did not effectively discharge solids even at 2 ex/hr. During these trials, the radial flow transported the settleable solids to the center portion of the tank, but water velocities were so low in the middle of the tank that a good portion of these solids settled within in a near region about the center drain (Summerfelt et al., 2000b). Accumulated settled solids are usually near to the center drain so that pulling an external stand-pipe regulating the bottom-center drain flow, even for an interval of < 1 min, will usually create sufficient flow to flush these accumulated solids. The presence of fish will improve solids removal in this area (Summerfelt et al., 2000b).

The radial flow mechanism played a much larger role than the mass transport mechanism in the transport of solids to the bottom-center drain. However, the inlet flow and hence the overall HRT for the tank also plays a role in that some minimum HRT must be maintained in order to produce a radial force significant enough to move solids to the center

(Summerfelt et al., 2000b). As mentioned above, one exchange per hour was not effective in removing solids, although the deeper tanks were more successful at this rate than were shallower tanks (diameter depth ratio < 6).

Quite clearly, as the tank depth increases (or diameter to depth ratio decreases), the enrichment factor is much higher for the "deeper" tanks (Summerfelt et al., 2000b). This also indicates that previous recommendations (Larmoyeux et al. 1973) to use diameter depth ratios between 5 and 10 should be revisited according to these results. It appears that a diameter depth ratio less than 5 and perhaps as low as 2 may be preferred to shallower tanks based upon the increase in enrichment as characterized by the k value and $k/(Q/V_{tank} + k)$. This is very important because it definitely impacts the quantity of fish that can be maintained as a standing biomass per unit floor area. Fish behavioral characteristics must also be considered, e.g., flounder that need floor space versus tilapia that distribute well vertically in the water column.

4.8 RACEWAYS

Raceways have primarily been associated with the culture of salmonids. Continued attempts have been made to adapt raceway culture methods to RAS, with mixed results. Proponents of raceways will generally argue for raceway design due to their better utilization of floor space and easier handling and sorting of fish. Their disadvantage is their inability to rapidly self-clean under typical operating conditions, which increases the dissolution of nutrients and cBOD into the flow. Figure 4.12 shows the all too frequent task associated with maintaining raceways, that being multiple weekly or even daily cleaning of the settling areas that must be placed somewhere in the raceway. Also notice in this figure that the worker is cleaning the bars that keep the fish confined in the raceway. This accurately demonstrates that every solid surface screening material in particular will need frequent cleaning maintenance. Avoid placing materials in fish tanks whenever possible.

Figure 4.11 Some of the solids swept from a raceway's fish rearing area can settle in quiescent zones. Captured solids must be manually removed, as pictured here at the Pennsylvania Fish and Boat Commission, Big Spring Fish Culture Station, Newville, PA (from Summerfelt et al., 2000a).

Raceways are the most common rearing tank design being used in locations where aquaculture has tapped into huge groundwater resources (IDEQ, 1998; Summerfelt et al., 2000a), such as the Thousand Springs area in Idaho where large amounts of rainbow trout are produced. In Idaho, raceway dimensions are typically about 3–5.5 m wide, 24–46 m long, and 0.8–1.1 m deep (IDEQ, 1998). Raceways usually have a length to width ratio of 1:10 and a depth < 1.0 m, and require a high water exchange rate, e.g., one tank volume exchange every 10 to 15 minutes (Summerfelt et al., 2000a).

Raceways depend upon this high turnover rate to maintain sufficient water quality for fish growth. Water enters the raceway at one end and flows through the raceway in a plug-flow manner, with minimal back mixing. As a result, the best water quality exists at the head of the tank where the water enters, and then deteriorates steadily along the axis of the raceway towards the outlet. It is not be uncommon to see fish crowded towards the inlet end of the raceway in order to obtain oxygen where it is highest, or at the outlet end of a raceway to avoid high concentrations some other accumulating contaminant. During feeding activities, fish will also crowd to the feeders, since it is also more difficult to distribute feed throughout raceways than it is in circular tanks.

Velocity through the raceway will generally be between 2-4 cm/s, which is insufficient to effectively remove settled solids from the rearing space. Thus, frequent maintenance for cleaning of raceways is required. A variety of mechanisms can be used to increase solids removal, such as

placing periodic baffles across the raceway to artificially increase velocity (20 to 40 cm/s or 10 times the raceway average water velocity) and sweeping action. However, these baffles must be moved to work the fish, and also provide a substrate for biosolids growth in warmer temperatures. In practice, the management of raceways is based on their oxygen design requirements, rather than their cleaning requirements. The velocity required to flush solids from unbaffled raceways is much greater than the velocity required to supply the oxygen needs of the fish. In practical terms, raceways are incapable of producing the optimum water velocities recommended for fish health, muscle tone, and respiration.

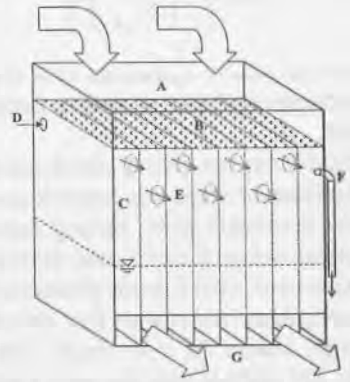


Figure 4.12 Low Head Oxygen (LHO) units are popular in raceway culture to restore oxygen in serial reuse raceway.

Since the difference between flow rates necessary to achieve a cleaning velocity and required flows for oxygen supply are so different, designs are based upon minimizing cross-sectional area to promote maximum water velocity. As a result, many raceway systems are operated in series, with the discharge of the upstream raceway serving as the inflow water of the next one downstream. Water may be treated between raceway sections to restore a particular water quality parameter for subsequent downstream utilization or removal, e.g., oxygen addition, CO_2 removal. Low head oxygenators (LHO's) have become popular in such applications (Watten, 1989). LHO's vary in configuration, but all are fundamentally similar in operation. These units consist of a distribution plate positioned over multiple (5 to 10) rectangular chambers, Fig. 4.12.

Water flows over the dam boards at the end of a raceway or is pumped upwards from an indoor fish tank, through the distribution plate, and then falls through the rectangular chambers. These chambers provide the gas-liquid interface needed for mixing and gas transfer. The streams of falling water impact a collection pool at the bottom of each chamber where the effluent water flows away from each chamber equally in parallel. Pure oxygen is introduced into the outer or first rectangular chamber, passes through the series of individual chambers, and finally is vented to the atmosphere at a much lower concentration. Each of the rectangular chambers is gas tight and the single holes between the chambers are properly sized and located to reduce back-mixing between chambers (Watten, 1989). These devices are completely described in Chapter 10 (Gas Transfer) and a computer program to predict their performance is given in the appendix and on the accompanying CD.

Figure 4.13 Raceways are often constructed side-by-side, with common walls. The raceway is slightly sloped, so that each raceway can be broken into several sections and the water serially reused as it flows from section to section.

Raceways can be constructed side-by-side, with common walls, for maximizing the utilization of floor space and reducing construction costs, Fig. 4.13. However, when constructed without common walls, because of their large aspect ratio (L:W), raceways require 1.5 to 2.0 times as much wall length as circular tanks. Circular tanks can also better structurally handle the weight of the confined water and can thus use thinner walls than rectangular tanks.

A quiescent zone devoid of fish is usually placed at the end of a raceway tank to collect the settleable solids that are swept out of the fish rearing area (IDEQ, 1998). At many large trout farms, these solids collection zones are the primary means for solids removal to meet discharge permit requirements (IDEQ, 1998). The maintenance of these settling zones can often be 25% of the total labor required for the entire fish operation (IDEQ, 1998).

MIXED-CELL RACEWAY TANKS

It should be fairly evident to the reader at this point that we strongly favor circular tanks because of their advantages of elevated water velocities, uniform water quality, and good solids removal characteristics. Still, the authors acknowledge that raceways make better utilization of floor space and allow easier handling and sorting of fish as compared to traditional, circular tanks. Their primary disadvantages are the large volume of water required, high turnover rates, and limited self-cleaning ability (Timmons et al. 1998). In practical terms, raceways are incapable of producing the optimum water velocities recommended for fish health, muscle tone, and respiration (Timmons et al. 1998). Even using lower exchange rates and lower velocities, use of raceways is being severely limited due to the unavailability of large quantities of high quality water, increased concern about their environmental impacts on receiving waters, and the difficulty in treating large flows from effluent discharge.

One solution to this problem is the cross-flow tank, a recent hybrid design that incorporates the desirable characteristics of both circular tanks and linear raceways (Watten and Johnson, 1990). Water is distributed uniformly along one side of a cross-flow tank via a submerged manifold and is collected in a submerged perforated drain line running the length of the opposite side.

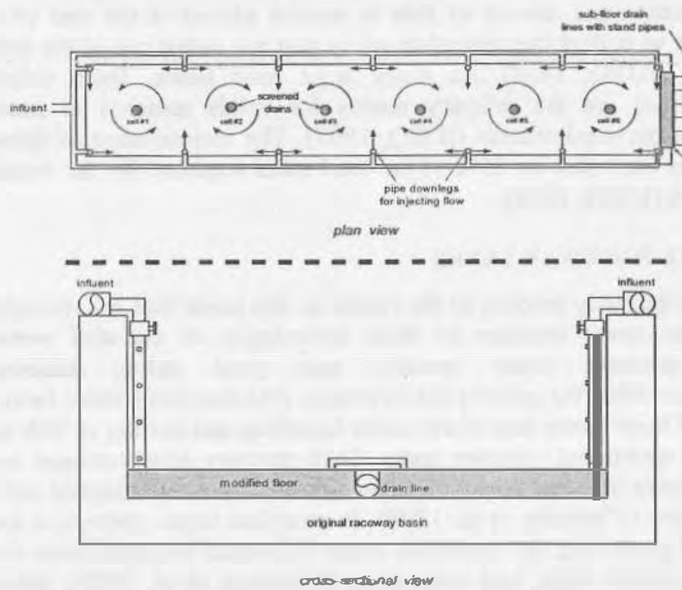


Figure 4.14 Illustration of a mixed-cell raceway tank (Watten, et al. 2000).

A second solution is to convert a raceway into a series of counter-rotating mixed-cells (Watten et al. 2000). The concept of a mixed-cell raceway was first proposed by Watten et al. (2000) to eliminate metabolite concentration gradients, increase current velocities, and improve solids scour at low water exchange rates. Watten modified a standard raceway section 14.5 m long and 2.4 m wide (Fig. 4.14) by creating a series of six counter-rotating cells, each 2.4 m by 2.4 m. A series of vertical pipe sections with jet ports were installed in the corners of each cell and water directed tangentially to create rotary circulation. The pipe sections can be swung up and out of the water during fish crowding or grading operations. Water was withdrawn from centrally located floor drain.

The basis for this design is that the influent is jetted perpendicular to the tank wall with sufficient force to establish a rotary circulation about the center drains in each cell. As a result, the standard raceway section is modified to create a series of horizontal counter rotating mixed cells with cell length equal to vessel width, Fig. 4.14.

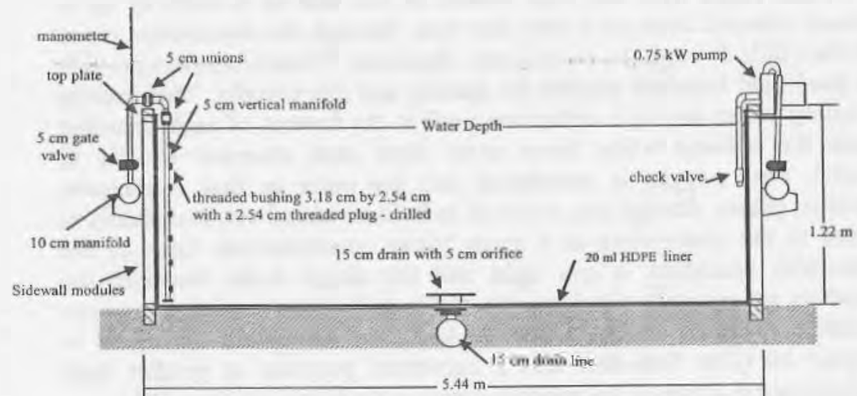


Figure 4.15. Cross-section of mixed-cell raceway showing construction details, pipe manifolds, vertical manifolds and drain details (Ebeling et al., 2005).

A prototype raceway was constructed in an existing greenhouse at the Conservation Funds Freshwater Institute with approximate dimensions of 16.3 m x 5.44 m x 1.22 m (54 ft x18 ft x 4 ft), which created three mixed-cells (Ebeling, et al. 2005). The basic design concept was to operate the raceway as a series of square/octagonal tanks, each having a center drain for continuous removal of solids and sludge. Each cell received water from four vertical manifolds (downlegs) extending to the raceway floor and located in the corners of each cell and at the intersection between adjacent cells (Figs. 4.15 and 4.16); four of the manifolds supply water to two cells concurrently. Water is pumped through several orifice discharges (or jet ports) from each of the downleg pipes to establish rotary circulation in the cell, with adjacent cells rotating in opposite directions. Each cell had a bottom drain located at the center of the cell connected to a drain line, which discharged solids and sludge to a settling sump.

This was then combined with the concept of the 'Cornell double drain system', where 10% to 20% of the total flow into a tank was removed from a center bottom drains and 80 to 90% of the flow was removed from the side drains (Timmons et al. 1998). Settable wastes and sludge were then removed from the center drains and collected in a settling sump.

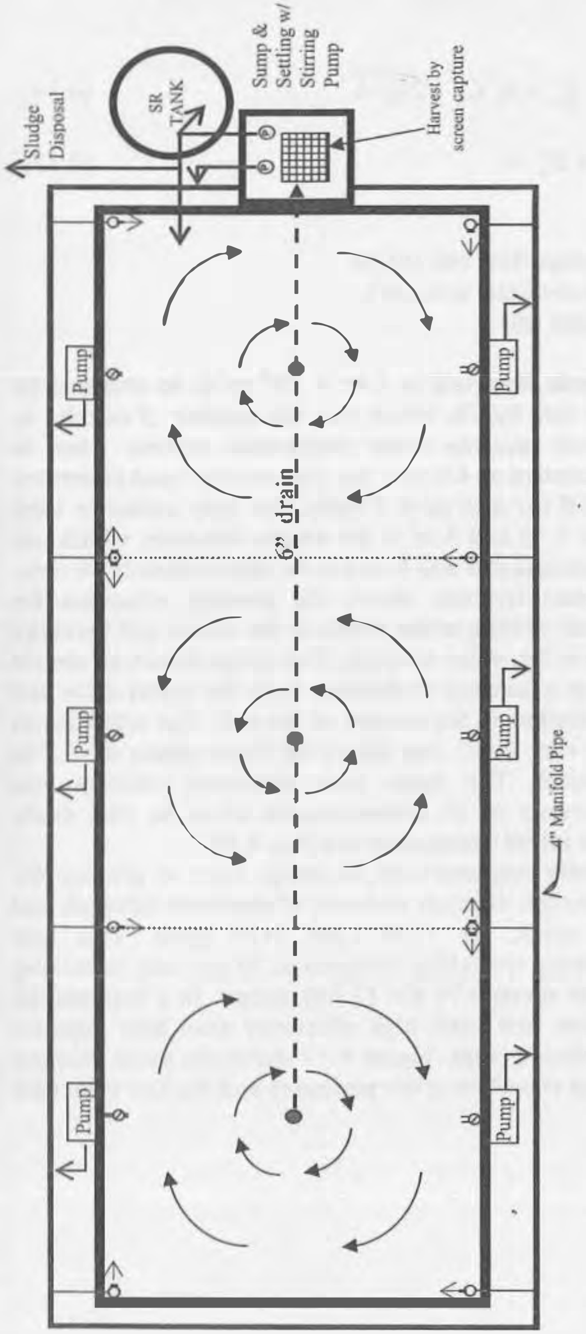


Figure 4.16 Mixed-cell raceway layout and flow pattern (Ebeling et al., 2005).

Labatut et al. (2006) conducted a series of experimental trials to evaluate the effect of nozzle diameter and the rate of bottom-center drain discharge on both the magnitude and uniformity of rotational velocities in the mixed-cell. Three nozzle diameters, 10, 15, and 20 mm, and three bottom-center flows, 0, 15, and 20%, were evaluated. Measurements of rotational velocities in the mixed-cell were made at 5 cm from the bottom of the tank. While the nozzle diameter was found to have a highly significant influence ($p < 0.01$) on the magnitude of the rotational velocities, the percentage of bottom flow did not ($p > 0.05$). Also, results suggested that uniformity of rotational velocities in terms of the radial-wise profile is not affected by either the nozzle diameter or the percentage of bottom flow.

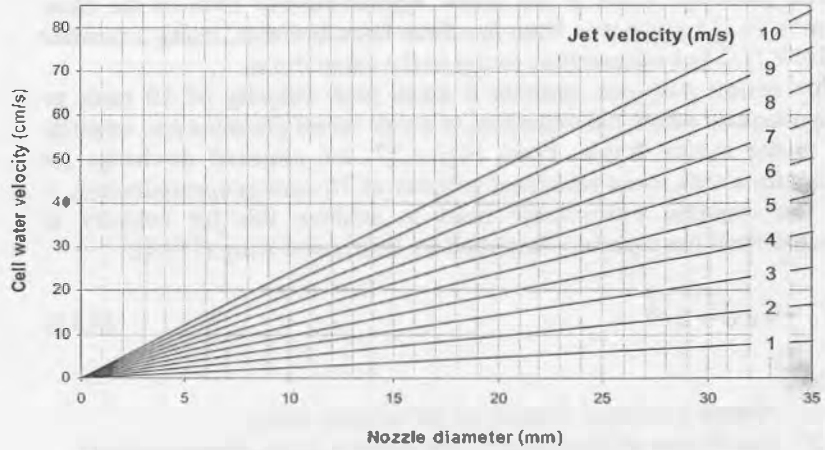


Figure 4.17 Iso-curves for predicting mean rotational velocities for different nozzle diameters and discharge jet velocities. Plot was based on bottom rotational velocities obtained in bottom flow rates that varied from 0 to 20%, a water depth of 1.15 m, and a mixed-cell diameter-to-depth ratio of 4.8:1. (From Labatut et al., 2007)

Labatut et al. (2007) also showed that the flux of momentum is the driving force controlling rotational velocities in a jet-forced circulation vessel and therefore jet velocity and nozzle diameter become the main variables to control. They found that the linear influence of the jet velocity on rotational velocities reported in previous studies remained valid provided that the nozzle diameter was maintained constant. Results of the study indicated that rotational velocities in mixed-cells follow a logarithmic trend as a function of the nozzle diameter for a constant jet

velocity. The linear and logarithmic models were combined to construct a set of iso-curves to predict rotational velocities as a function of jet velocity and nozzle diameter (Fig. 4.17). The iso-curves can be used to facilitate the design of a Mixed-cell Raceway where particular rotational velocities are desired.

As a design example, at an exchange rate of approximately 0.5 tank volume/hr, a flow rate of only 0.74 m^3/min (250 gpm) is required for the raceway in this example. This was accomplished using two 0.95 kW (1 hp) pumps which removed water at the two ends of the raceway and injected it into a 7.5 cm (3 in) manifold that circled the top of the raceway. Water was withdrawn either from a 5 cm (2 in) PVC pipe inlet located approximately 25 cm below the surface at one end or an end sidewall discharge drain at the other. Approximately 15% of the flow 0.13 m^3/min (35 gpm) was from the three bottom drains, using a smaller 0.375 kW (1/2 hp) submersible pump in the sump drain.

For design purposes, assume a mean tank velocity of 10 cm/s to insure adequate rotational velocities to move waste particles and uneaten food to the center drains. From Fig. 4.17, the required discharge jet velocity for a tank mean rotational velocity of 10 cm/s is approximately 4 m/s. The required piezometric head to achieve this jet velocity is computed from the equation described by Brater and King (1976):

$$V_o = C_d \sqrt{2 \cdot g \cdot h} \tag{4.13}$$

where,

V_o Nozzle discharge velocity or jet velocity (m/s)
C_d Coefficient of Discharge of the nozzles (0.93, dimensionless)
g Acceleration due to gravity (9.81 m/s^2)
h Piezometric head, i.e., pressure head upstream of the nozzle (m)

The C_d of the nozzles was obtained from a series of flow rate measurements in four jet port manifolds at different piezometric heads. Details of the experiment and data are given by Labatut (2005). The average C_d reported by Labatut (2005) was consistent with the values found in literature for this kind of entrance (Brater and King, 1976). By using Equation (4.13), a jet velocity of 4.0 m/s, and a C_d of 0.93, the required piezometric head was calculated to be 1.0 m.

A requirement of the system design was to maintain a water exchange rate of 0.5 volumes per hour, i.e., a total system flow rate of 0.74 m^3/h. In order to keep a constant jet velocity and flow rate, Equation 4.13 can be modified to include the nozzle flow rate and nozzle cross-sectional area and solve for the required nozzle diameter:

$$Q_o = A_o \, C_d \sqrt{2 \cdot g \cdot h} \tag{4.14}$$

with

$$A_o = \pi \, D_o^2 / 4 \tag{4.15}$$

where,

Q_o Nozzle discharge flow rate (m^3/s)
A_o Nozzle cross-sectional area (m^2)
D_o Nozzle diameter (m)

The calculated nozzle flow rate is 1.48×10^{-2} m^3/s, as obtained by dividing the total flow rate by 50, which was the number of nozzles in the prototype mixed-cell raceway water distribution system. Also, to keep the jet velocity constant at 4.0 m/s, the piezometric head computed in the previous step (1.0 m) was used. Finally, the only unknown term remaining in Equations 4.14 and 4.15 is the nozzle diameter, which can then be explicitly determined and was found to be approximately 10 mm.

Figure 4.18 (10 mm orifices) shows the average velocities for annular rings 0.5 m wide starting at the center in the mixed cell (average values for three depths in the water column). This graph shows an almost linear velocity profile as a function of distance from the center drain and significant cleaning velocities in the corners of the cell. The velocities in vertical direction were very small, just above the drain values of -2.2 to 2.8 cm/s were measured. The mean tank rotational velocity was estimated from the average of all measurements taken as 10.5 cm/s, consistent with both the initial assumption and Fig. 4.18.

High biomass density requires high exchange rates to provide for significant dissolved oxygen demand, removal of ammonia-nitrogen and carbon dioxide and solids, i.e. 1700 Lpm (450 gpm). This was accomplished by increasing the orifice diameter to 20 mm and increasing the number of pumps to seven 0.74 kW (1 hp) pumps. In a commercial application, two, or three, low head, high efficiency axial flow impeller pumps, would replace these pumps. Figure 4.19 shows the mean velocity profile the high cleaning velocities at the perimeter and the low velocities near the center drain.

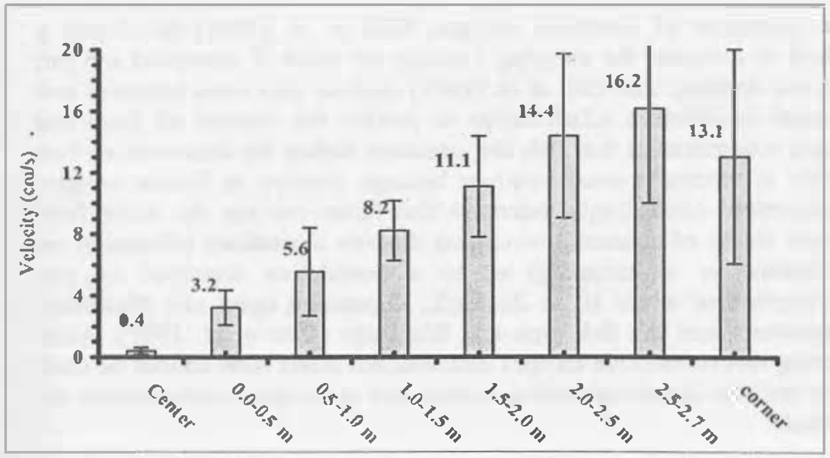


Figure 4.18. Cell #3 mean velocity profile with 10 mm orifice diameter, 100 cm pressure head, recirculating 0.74 m^3/min (250 gpm) or approximately 0.5 tank exchanges/hr and 15% withdrawal from the center drains (from Labatut et al., 2007).

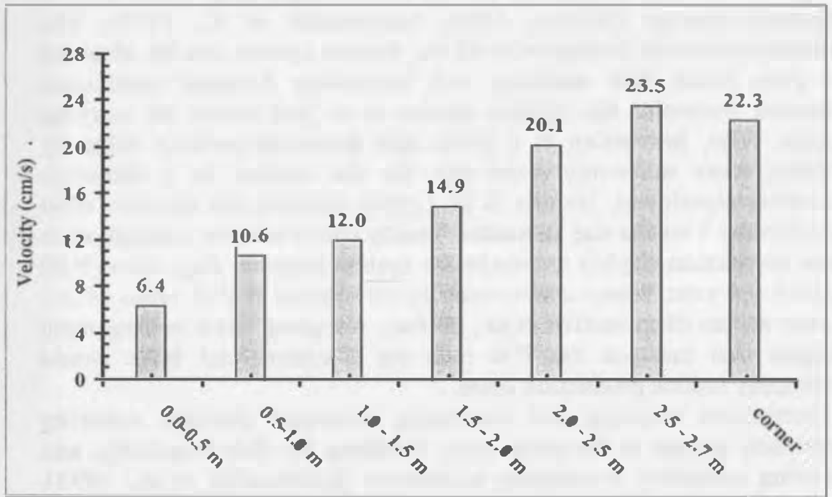


Figure 4.19. Cell #3 mean velocity profile with 20 mm orifice diameter, 135 cm pressure head, recirculating 1700 Lpm (450 gpm) or approximately 2-tank exchanges/hr and 15% withdrawal from the center drains (from Labatut et al., 2007).

Raceways have several inherent advantages over circular tanks, including ease of sorting, grading, and handling fish and optimization of floor space. Results of these studies showed excellent bottom velocities for scouring solids and moving them towards the center drains in each cell of the prototype mixed-cell raceway at several different exchange rates. Mixed-cell raceways show excellent potential for either retrofitting existing raceways or as a design for a new production system

4.9 Carrying Capacity Issues

Production efficiency in culture tanks can be boosted by increasing the culture tank's carrying capacity, which in simplistic terms is the maximum fish biomass that can be supported at a given feeding rate. Dissolved oxygen is usually the first water quality parameter to limit culture tank carrying capacity, because the fish consume dissolved oxygen during respiration. The amount of dissolved oxygen available in fish culture tanks is dependent upon the water flow rate multiplied by its concentration of available dissolved oxygen, i.e., the inlet dissolved oxygen concentration minus the minimum allowable dissolved oxygen concentration, assuming no in-tank aeration or significant photosynthesis. For example, assuming that water is not limiting and that its dissolved oxygen concentration is constant, doubling the water flow, i.e., hydraulic exchange rate, through a culture tank will double the carrying capacity of the tank. However, moving more water through the culture volume is expensive, as it requires large pumps, pipes, or water pressure requirements, which may not be feasible, and will increase the farm's fixed and variable costs. Alternatively, supersaturating the dissolved oxygen in the water flowing into the tanks has also been popular, and often is a more cost effective method to improve the profitability of tank-based fish farms (Colt et al. 1991). For example, assuming a minimum allowable outlet dissolved oxygen concentration of 7 mg/L, increasing the dissolved oxygen concentration entering a culture tank from 10 mg/L to 16 mg/L would triple the culture tank's available oxygen and thus triple its carrying capacity (Summerfelt et al., 2000a).

Supersaturating the flow with dissolved oxygen can be achieved cost effectively with many different oxygen transfer devices, even in low head applications (Boyd and Watten, 1989). The cost of adding oxygen at a commercial tank-based fish farm can be estimated from the following farm and species specific variables:

- oxygen consumption rate per unit feed consumed, e.g., 0.25 kg oxygen per kg feed

- feed conversion rate, e.g., 1.3 kg of feed is required to produce 1.0 kg of fish
- transfer efficiency of oxygen gas into water, e.g., 1.0 kg of oxygen feed-gas must be supplied to dissolve 0.6 kg oxygen (as an example)
- density of oxygen gas, e.g., 0.75 cubic meter oxygen gas (at 20°C and 1 atm pressure) per kg oxygen
- cost of oxygen gas, e.g., $0.15 per cubic meter oxygen gas (at 20°C and 1 atm pressure)

As an example, estimates for each of the variables listed above are used to make a simplistic prediction of the cost of oxygenation, assuming all dissolved oxygen requirements are provided by the purified oxygen feed gas:

$$oxygen\ \text{cost} = \frac{1.3\,kg_{feed}}{1.0\,kg_{fish}} \cdot \frac{0.25\,kg\ O_2}{kg_{feed}} \cdot \frac{1.0\,kg\ O_2\ Supplied}{0.6\,kg\ O_2} \cdot \frac{0.75\,m^3\ O_{2\ gas}}{kg\ O_2\ Supplied} \cdot \frac{\$0.15}{m^3\ O_{2\ gas}}$$

$$= \frac{\$0.061}{kg_{fish}}$$

In this example (the 0.6 kg O_2 factor epresents a 60% efficiency on transfer efficiency), the cost of the oxygen feed gas required to produce 1.0 kg fish would amount to $0.061. At this same feed conversion rate, fish feed alone might cost more than $0.50 for every kg of fish produced. Therefore, the cost of the oxygen would be relatively low, much lower than the cost of the required fish feed. Thus, adding a pure oxygen transfer system to increase fish farm carrying capacity can be used to decrease production costs by increasing fish production in the same culture tanks while using relatively similar labor requirements.

Other fish metabolites such as dissolved carbon dioxide, ammonia, and suspended solids will limit culture tank carrying capacity, after oxygen is no longer limiting. Fish can produce roughly 1.0–1.4 mg/L total ammonia nitrogen, 13–14 mg/L dissolved carbon dioxide, and 10–20 mg/L of total suspended solids (TSS) for every 10 mg/L of dissolved oxygen that they consume. Without some form of ammonia or carbon dioxide control, dissolved carbon dioxide and un-ionized ammonia concentrations can rapidly accumulate to toxic levels when fish consume large quantities of dissolved oxygen. Colt et al. (1991) developed a method to estimate the carrying capacity of water if dissolved oxygen were not limiting. The Colt et al. (1991) method uses mass balances and chemical equilibrium relationships to predict the amount of dissolved oxygen concentration that fish can consume before the dissolved carbon dioxide or ammonia concentrations become limiting to further oxygen consumption. Accordingly, intensive fish farms can use the water flow without worry of ammonia or carbon dioxide limitations (assuming no biofiltration or air-stripping) up to a cumulative dissolved oxygen consumption of about 10 to 22 mg/L, depending upon pH, alkalinity, temperature, and the fish type and life stage (Colt et al. 1991). After reaching this cumulative oxygen demand, the water flow cannot be used again until the dissolved carbon dioxide and ammonia concentrations are reduced.

4.10 Stock Management Issues

Fish farm production can also be increased (approximately doubled) through the use of a continuous production strategy, rather than a batch production strategy (Watten, 1992; Summerfelt et al., 1993). The maximum economic productivity of the culture system can be obtained with year round fish stocking and harvesting because continuous production maintains the culture system at or just below its carrying capacity. Also, harvesting at a given size increases product value by providing more uniformly sized fish for the market. In a full-scale production experiment, Heinen et al. (1996) showed that rainbow trout stocked every 8 weeks and harvested weekly could achieve a steady-state annual production (kg/yr) to maximum system biomass (kg) ratio (P:B) of 4.65:1 per year. Many commercial farms operate at P:B ratios of 3:1 per year or less (Summerfelt et al., 2000a). Adopting stock management strategies that increase the P:B ratio on a commercial farm would significantly reduce production costs.

Continuous stocking and harvesting strategies requires culturing several size groups at the same time, handling the fish frequently, and improving inventory accounting techniques (Summerfelt et al., 1993). This can stress the fish and increase labor costs if automated equipment is not used. To keep the cost of handling and grading fish to a minimum, and the stress on the fish to a minimum, a convenient mechanism for size sorting the fish, counting them, and moving them to other locations should be incorporated into the culture tank and facility design (Summerfelt, 2002). Simply netting the fish out of the tank, or using a

net to crowd the fish for harvest or grading is an obvious solution. More sophisticated crowding and grading can be achieved using crowder and grader frames or gates that move down the length of a raceway or pivot around the center of a circular culture tank (Piper et al., 1982; Summerfelt, 2002; Summerfelt et al., 2004a). Fish sufficiently small in size can swim through the grader bars while the larger fish are retained behind the gate. Use of crowder and grader gates is thought to be less stressful to the fish, because their use does not require handling the fish or moving them out of the water (Summerfelt, 2002; Summerfelt et al., 2004a). And once crowded, fish can also be induced to swim through channels, pipes, or raceways to another location with relatively little stress. Crowded fish can also be moved rapidly to other areas using more aggressive yet relatively safe fish pumps, or brail nets and cages when they are used with care (Summerfelt, 2002; Summerfelt et al., 2004a; Clingerman et al., 2005).

Hand grading devices such as box graders are common at many hatcheries and have proven to work well on small farms, where the cost of automatic grading and counting equipment cannot be justified. At larger farms, however, labor and fish stress can be reduced with the wise use of automated grading and inventory tracking equipment. Commonly used automatic graders include mechanically driven belt graders and roller graders. These mechanical graders usually require removing the fish from water for a brief period as they pass through the sizing mechanism. Although mechanical graders produce some stress and trauma, many of the established commercial mechanical grading machines are considered safe, reliable, and fast methods to size sort and count large numbers of fish. However, as with all new technologies, before purchasing equipment of this type, it is best to check with other fish farmers who have used the equipment. Other interesting and new technologies that use ultrasonic, infrared light and video systems are now commercially available to estimate fish size distributions within culture tanks and cages (Summerfelt, 2002). This type of inventory tracking equipment can sometimes be used to track fish growth, feed conversion (if feed input and fish numbers are known), and estimate the fraction of fish reaching harvest size.

4.11 Scale Issues

The number and size of culture tanks is an important factor to be considered during the design of the fish farm and its stock management plan (Timmons et al., 1998; Summerfelt et al., 2000a). It is now

becoming a more common practice for fish farms to use fewer but relatively larger culture tanks to meet culture volume requirements. For example, Karlsen (1993) described how more recent land-based salmon-smolt farms in Norway have reduced the number of tanks to between six to eight units, much fewer in comparison to earlier farms. No matter where the fish farm is located or what species the farm is producing, it often becomes apparent that fewer (maybe six to ten) but relatively larger culture tanks can provide culture volume much more cost effectively than using many (perhaps 30 to 100) correspondingly smaller tanks. Also, the costs of miscellaneous equipment and labor decrease when a given culture volume can be achieved with a few larger culture tanks rather than with many smaller tanks. Use of fewer but larger tanks reduces the purchase and maintenance of feeders, dissolved oxygen probes, level switches, flow meters/switches, flow control valves, and effluent stand-pipe structures (Timmons et al. 1998). Use of fewer but larger tanks also reduces the time required to analyze water quality, distribute feed, and perform cleaning chores, i.e., the times are about the same for a larger tank as for a smaller tank, (Timmons et al. 1998). Also, the time and logistics of fish management in a large number of tanks can become quite costly.

However, the advantages achieved through the use of larger tanks must be balanced against the risk of larger economic loss if a tank fails due to mechanical or biological reasons (Summerfelt and Bebak-Williams, 2006). There are also difficulties that could arise in larger culture tanks when removing mortalities, grading and harvesting fish, and controlling flow hydraulics, e.g., water velocities, tank mixing, dead-spaces, and settling zones. Therefore, large culture tanks must be designed properly to allow for fish management and control over flow hydraulics, as discussed elsewhere in this chapter.

4.12 Mechanisms To Remove Dead Fish

Daily removal of dead fish from the bottom center drain is important. Dead fish in the fish culture system negatively influence profits, fish health, water quality, solids removal, and the water level in the tank. Commercial fish farmers need a simple and reliable way to remove daily mortalities with minimal labor. There are varieties of approaches to address this task. When applicable, an uncovered bottom center drain makes the task of removing dead fish easy. When dead fish sink, they are carried in the radial flow to the bottom center drain where they are sucked through to the external standpipe chamber.

Methods to remove dead fish and/or waste solids from the bottoms of deep culture environments have been developed for large floating cages (Braaten, 1991), which when enclosed in bags rather than nets look very similar to circular culture tanks. These mortality and solids removal methods may be transferable to land-based circular culture tanks.

The Freshwater Institute has been working to develop practical, efficient, and labor reducing dead fish collectors that can be incorporated into the dual-drain particle trap mechanism, Figs. 4.21 and 4.22. The more basic method for removing dead fish incorporates the center drain outlet screen into the inner pipe of a two-pipe center post system, Fig. 4.21. The outer pipe consists of a post secured in the tank floor such that large openings cut into the pipe just above the tank floor level allow dead fish to pass through the outer pipe when the inner pipe is lifted. The sizes of the outer and inner concentric pipes have been selected to make a close but unhindered fit. To conveniently flush dead fish captured at the bottom center drain, the inner pipe is raised inside of the fixed center post while the external standpipe over the mortality drain is removed; this produces a surge of flow that carries the dead fish out of the tank. This mortality removing mechanism worked well but, because it requires placing a center post in the tank, the authors prefer a more sophisticated approach that uses a pneumatic piston to lift the center screen out of the way to flush dead fish out of the tank, Fig. 4.20. The pneumatic mort flushing system has worked well and does not block the center of the tank when harvesting devices are used. Both mort flushing systems are described in detail in Summerfelt (2002).

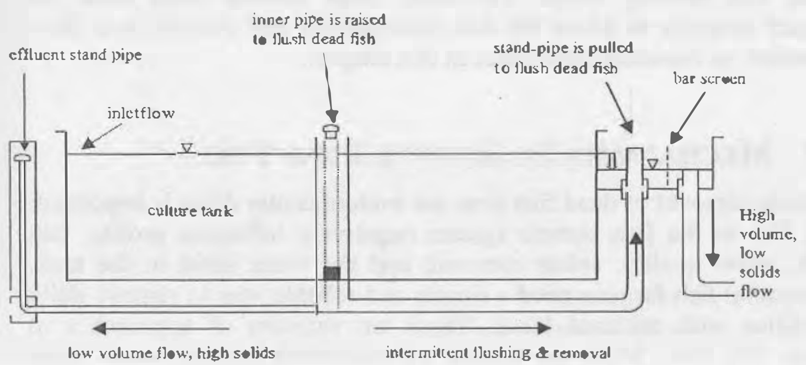


Figure 4.20 A concentric pipe system to flush solids and remove dead fish from the bottom center drain; elevated drain for removing the high volume, low solids effluent (from Timmons et al., 1998).

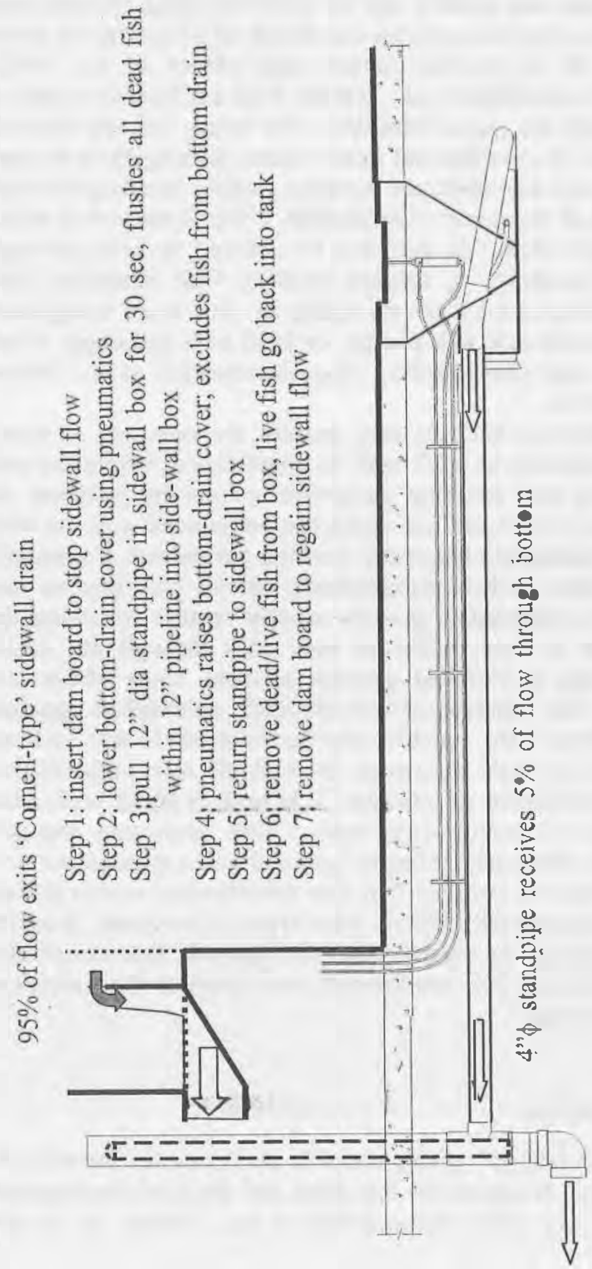


Figure 4.21 Pneumatic mort flushing system at the Freshwater Institute, shown in normal operating mode (from Summerfelt et al., 2004a).

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LIST OF SYMBOLS

BL	Fish body length, dimensionless
C	Concentration of solids in tank water, kg/m^3
$C_{density}$	1.5 for L in inches (0.24 for L in cm) for tilapia
$C_{out,b}$	Concentration of pellets leaving bottom center drain, kg/m^3
COC	Cumulative oxygen consumption, kg
$D_{density}$	Carrying capacity or biomass density, lb/ft^3 (kg/m^3)
DDR	Diameter depth ratio of tank, dimensionless
DO	Dissolved oxygen, mg/L
g	Acceleration due to gravity, m^2/s (ft^2/s)
H	Dynamic heads ($v^2/2g$), m (ft)
HRT	Hydraulic residence time, 1/h
k	1st order rate constant characterizing bead enrichment at the bottom-center drain
L	Fish length, inches (cm)
$L_{raceway}$	Length of the raceway in meters
MRT_{ideal}	Ideal mean residence time, 1/h
P:B	Ratio of yearly production to biomass carrying capacity
Q	Inlet flowrate, m^3/s
$Q_{out,b}$	Flowrate flushed through the bottom center drain, m^3/h
R	Water exchange rate in a raceway design, volumes/h
t	Time
V_{orif}	Velocity through the water inlet structure (orifices or slots), m/s
$V_{raceway}$	Raceway velocity, cm/sec
V_{rota}	Rotational velocity in the tank, m/s
V_{safe}	Fish body lengths per second, BL/s
V_{tank}	Water volume in the tank, m^3 or L
α	Proportionality constant to relate tank and orifice velocity
φ_{enrich}	Relative enrichment factor due to Cornell dual drain, %
ρ	Density of water, kg/m^3

CHAPTER 5

SOLIDS CAPTURE[1]

5.0 OVERVIEW

Suspended solids adversely impact all aspects of a recirculating aquaculture system (RAS), so the first objective of any recirculating treatment scheme is the removal of solid wastes. As aquaculture systems intensify, the management of solids, through feed design, feeding management and flow regulation and eventual removal using separation and sludge treatment technology becomes increasingly important (Cripps and Bergheim, 2000). Suspended solids are generated from feces, biofloc (dead and living bacteria), and uneaten food. These suspended particles will vary greatly in size from centimeters (cm) to microns (µm). Aquacultural solids are characterized by size into classes, as shown in Fig. 5.1 (Vinci et al. 2001). The term "fine solids" is used herein to identify the solid particulates that do not readily settle from the water column, e.g., < 100 µm. As a prelude to understanding the descriptions of size distribution and contribution by weight of the various size classes, it is important to know that samples are usually pre-filtered to remove large particles, i.e., >200 µm. The contribution of these large solids to TSS measurements is added to the solids concentration obtained from the filtered water when TSS is reported for a system's performance characterization. In practical applications, these larger particles should always be removed first and must be a primary focus, since if they are not removed, they become "smaller" more difficult particles to remove.

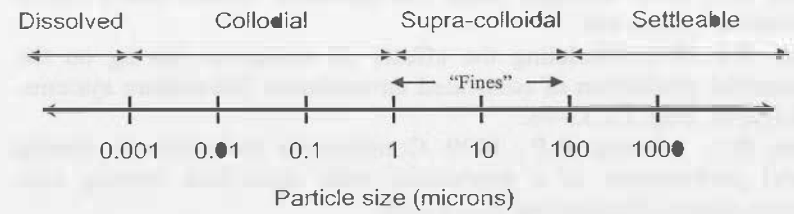

Figure 5.1 Solid size characterizations in aquaculture waters.

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In RAS, the majority of particles by weight will be less than 100 μm in size; and in intensive RAS systems the majority of particles by weight will be 30 μm or less in size. In such cases, mechanical filtration will be ineffective. Chen et al. (1993b) showed that 80% to 90% of the total weight of solids in a tank (prefiltered to remove particles >130 μm; see Fig. 5.2) was comprised of individual solid particles that were 35 μm or less in diameter.

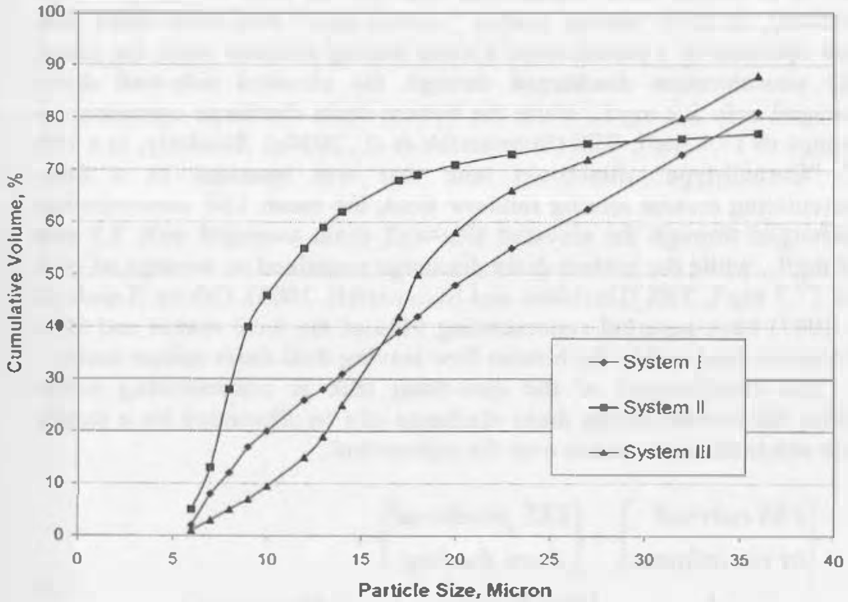


Figure 5.2 Cumulative distribution of particle volume in three recirculating high density aquaculture systems (Chen et al., 1993b). (Systems I and III were rainbow trout and System II was brook trout. Feeding rates were approximately 0.75% BW per day. Water exchange was 5% per day. System III used ozone.)

All sizes of particulates must be addressed and managed by an appropriately selected treatment method for particles within each size range, e.g., sedimentation and screening for removal of large particles and foam fractionation or ozone treatment for fine solids removal. Granular media filters can control a wide range of particle sizes, but can also promote particle dissolution if the solids are stored in the process flow for an extended duration (Chen et al., 1993a; Golz, 1999). This type of filter can be effective in removing solids down to about 20 μm, and is the favored choice of many designers for systems that have relatively low feed loading levels, high reuse, or high clarity demands. This and other solids removal techniques are reviewed in this chapter.

Total suspended solids (TSS[2]) concentration is defined as the mass of particles above 1 μm in diameter (APHA, 1995) occurring in a known volume of water. Suspended solids have both inorganic and organic components. The organic portion, known as volatile suspended solids (VSS), contributes to oxygen consumption and biofouling problems. The inorganic components (ash) contribute to formation of sludge deposits. Physically, suspended solids can be partitioned further into settleable solids, typically greater than 100 μm, and non-settleable suspended solids, which are less than 100 μm (EPA, 1975). The finer, non-settleable suspended solids are more difficult to control and cause most of the problems in recirculating systems.

Fine suspended solids are extremely detrimental to general fish health (Magor, 1988; Chapman et al. 1987; Alabaster and Lloyd, 1982). Gill function can be impaired by the accumulated solids smothering the gill and compromising oxygen transfer or by offering a suitable habitat for the proliferation of pathogenic organisms (Cripps and Bergheim, 2000; Liltved and Cripps, 1999; Braaten, 1992). Experts in the field have not yet agreed on a set of definitive design values for acceptable TSS concentrations, which would then serve as a system design goal for the TSS removal efficiency of RAS designs. For example, according to Alabaster and Lloyd (1982), for inland fisheries, there is no evidence that concentrations of suspended solids up to 25 mg/L have any harmful effect on fish. The EIFAC (1980) suggests that TSS concentrations be maintained below 15 mg/L as a safe value in recirculating systems, while Muir (1982) recommends a limit of 20 to 40 mg/L for these same systems. The authors have grown tilapia in systems that had TSS in excess of 100 mg/L and still maintained good fish productivity, but this was in an absence of virtually all other stressors. Keep in mind that different fish species may have significantly different tolerance levels to solids concentrations and that other water quality parameters may impair a fish's ability to withstand high TSS concentrations.

Finally, during this initial introduction section, we cannot overemphasize the importance of rapid and complete solids removal from the culture vessel. All other unit processes will fail if this primary function is poorly performed. The case history failures described in

[2] All acronyms, abbreviations, and variable symbols are defined at the end of the chapter.

Chapter 17 were primarily attributed to lack of effective solids removal from the culture vessels.

5.1 Solids Balance

Whenever possible, water flows should be managed to concentrate solids in a small portion of the total flow for the system. An effective way to do this is by using the Cornell Dual-Drain circular tank. In this system, 10% to 20% of the total flow exits the tank from a center bottom drain while the majority of the flow exits from the tank sidewall. (This concept is discussed in much more detail in Chapter 4: Culture Units). Use of the dual-drain approach greatly increases the concentration of solids being removed from the low flow bottom center drain. The concentration of solids in this low flow is typically 6-10 or more times higher than the concentration of solids that exit through the main flow drain (Twarowska et al., 1997; Davidson and Summerfelt, 2004; 2005; Summerfelt et al., 2004a), whether it is located in the tank sidewall or as an upper center drain.

As an example of the effectiveness of the dual-drain system, the author has measured the results achieved in an intensive tilapia system (daily feeding rates were approximately 80 kg/day; tank volume was 53 m^3). In this system, the concentration of suspended solids in the water exiting the side-wall drain was the same as the concentration of solids in the overall tank (6.4 mg/L, st. dev.=3.6 mg/L). This means that practically speaking the net solids removal from the tank were in fact removed by the center bottom drain flow, which exited the tank at a rate of 110 L/min. This center drain discharge flow of 110 L/min is in comparison to the total water flow of the system to the biofilters and water conditioning units of 3.6–5.5 m^3/min (Timmons, unpublished data). The net settleable solids that were removed through the center bottom drain were then subsequently captured by a mechanical screen filter and/or a settling tank (emptied daily; approximately 3 m^3). The authors have designed several systems that successfully use the Cornell dual-drain approach and the design has been widely adapted in the tilapia, sturgeon, salmon smolt (Forsythe and Hosler, 2002; Holder, 2002; Vinci et al., 2004), and Arctic char (Summerfelt et al., 2004b) farming industries.

> **"Rule of Thumb"**
>
> Cornell Dual-Drain can increase center drain TSS by 10-fold.

Research at the Freshwater Institute (Shepherdstown, WV) on solids removal within a "Cornell-type" dual-drain tank operated in a single-pass mode has shown that the mean total suspended solids (TSS) concentrations discharged through the elevated side-wall drain averaged only 1.5 mg/L, while the bottom drain discharge contained an average of 19.6 mg/L TSS (Summerfelt et al., 2000). The tank contained rainbow trout at a density of 60 kg/m^3 and was fed 1% body weight per day. Similarly, in three heavily loaded "Cornell-type" dual-drain tanks that were operated in a partial-reuse system rearing rainbow trout, the mean TSS concentration discharged through the elevated side-wall drain averaged only 2.2 mg/L, while the bottom drain discharge contained an average of 17.1 mg/L TSS (Summerfelt et al., 2004a). Similarly, in a 150 m^3 "Cornell-type" dual-drain tank that was operated in a fully recirculating system rearing rainbow trout, the mean TSS concentration discharged through the elevated side-wall drain averaged only 3.2 and 4.5 mg/L, while the bottom drain discharge contained an average of 16.5 and 27.7 mg/L TSS (Davidson and Summerfelt, 2005). Others (Lunde et al. 1997) have reported concentrating 91% of the fecal matter and 98% of uneaten feed within the bottom flow leaving dual-drain culture tanks.

The effectiveness of the dual-drain tank at concentrating solids within the bottom center drain discharge can be illustrated by a steady state solids balance written over the culture tank,

$$\begin{Bmatrix} TSS\ carried \\ in\ via\ \text{influent} \end{Bmatrix} + \begin{Bmatrix} TSS\ produced \\ from\ feeding \end{Bmatrix} = \begin{Bmatrix} TSS\ leaving \\ sidewall\ outlet \end{Bmatrix} + \begin{Bmatrix} TSS\ leaving \\ center\ drain \end{Bmatrix} \tag{5.1}$$

or more explicitly,

$$\{Q \cdot TSS_{in}\} + \{P_{TSS}\} = \{Q_{out1} \cdot TSS_{out1}\} + \{Q_{out2} \cdot TSS_{out2}\} \tag{5.2}$$

P_{TSS} is determined by using the following equation:

$$P_{TSS} = a_{TSS} \cdot r_{feed} \cdot \rho_{fish} \cdot V_{tank} \tag{5.3}$$

The fraction of solids removed (f_{rem}) through the tank center drain can be determined from the following equation:

$$f_{rem} = \frac{Q_{out2} \cdot TSS_{out2}}{(Q_{out1} \cdot TSS_{out1}) + (Q_{out2} \cdot TSS_{out2})} \tag{5.4}$$

Equation 5.3 can be rearranged, substituted into Eq. 5.2 and then rearranged again to allow for TSS_{out2} to be calculated from f_{rem}, Q, Q_{out2}, P_{TSS}, and TSS_{in}:

$$TSS_{out2} = \left(Q \cdot TSS_{in} + P_{TSS} \right) \frac{f_{rem}}{Q_{out2}} \tag{5.5}$$

5.2 Basic Design Parameters for Round Tanks

Most of this design description material is presented in Chapter 4 Culture Units, but is repeated here for convenience. Round tanks will operate as self-cleaning vessels, if the diameter-to-depth ratios are maintained within a recommended range. It is for this reason that we highly recommend round tank culture vessels. Round tank vessels should be designed using the following criteria:

- Use a tank diameter-to-depth ratio between 3 and 10 and preferably between 3 and 6. For example, if you are using a tank that is 2 m deep, then the acceptable range of diameters is from 6 m to 20 m (the 3 to 10 range for diameter to depth ratio) or a 1 m deep tank could be from 3 to 10 m in diameter.
- Employ the Cornell dual-drain design with the center drain sized to flush at least 6 L/min of flow per m^2 tank plan area (Davidson and Summerfelt, 2004), which generally will require 5% to 20% of the total flow used to operate the tank; remove the remaining percentage of flow, i.e., 80% to 95%, from the upper half of the outside tank wall (see Chapter 4).
- Maintain water velocities about the tank perimeter of at least 15 to 30 cm/s to promote the movement of solid wastes towards the center drain.

Once the larger solids have settled and been "flushed" from the culture tank, the next step is to remove the suspended solids from the water column before returning the water to the culture tank vessel. The

various methods for removal of suspended solids are discussed in the following paragraphs.

5.3 Solids Generation

Virtually all the wastes generated within a recirculating system originate from the feed. These manifest in two ways: a) uneaten feed and b) fish excrement in the form of solids, liquid, or gas. Of the feed that is eaten, 80% to 90% will eventually be excreted in some form (Hopkins and Manci, 1989). As a rule of thumb, use 25% of the quantity fed to the fish as the mass that will be produced as suspended solids (or total suspended solids, TSS) on a dry matter basis. TSS produced by fish is primarily in the form of feces. The mass production rate of feces is proportional to the feeding rate and is discussed in more detail in Chapter 3 Loading Rates and several literature references (Liao and Mayo, 1974; Wimberly, 1990; Malone et al. 1990; Westers, 1989; Iwana, 1991; Page and Andrews; 1974 Patterson et al., 1999; Patterson and Watts, 2003; Davidson and Summerfelt, 2005).

"Rule of Thumb"

TSS = 25% of Feed Fed
(dry matter basis).

5.4 TSS Physical Characteristics

From the perspective of solids control, the two most important physical characteristics of suspended solids in a recirculating system are:

- particle specific gravity
- particle size distribution

Specific gravity is determined by the source of the particles, while the size distribution is determined by a combination of factors, including the solids removal process, the source of particles, feed properties, fish size, water temperature, and turbulence in the system.

The behavior of suspended particles in water is determined by their particle size and specific gravity. Specific gravity is defined as the ratio of the density of a wet particle to that of water (APHA, 1989). Based on the measurements of solids taken from two recirculating systems, Chen et al. (1993b) reported that the average specific gravity of the particulate

matter was 1.19, somewhat greater than that of the water. Based on the author's findings, however, Chen's value is probably greater than should be used in filter design calculations, as the authors have found that a specific gravity of 1.05 is more representative of suspended particles originating as tilapia feces. This is perhaps due to the fecal casing that enshrouds tilapia feces, making it less dense. The point is that fish feces is not much "heavier" than water and therefore does not settle as rapidly as would aggregate material of the same size.

While feces are the source of most of the suspended solids, uneaten feed is also a significant source of TSS in fish culture water. TSS from feed typically has a different particle size distribution than the TSS originating as feces. Uneaten feed subsequently breaks down slightly in the water column, but even after several hours and repeated passage through pumps, over 97% of the feed particles will be greater than 60 µm in size and 73% will be larger than 500 µm (0.5 mm). The particles of suspended solids originating from these two sources (feces and uneaten feed) are notably different in size and specific gravity and therefore respond to control mechanisms in different ways. In RAS waters, fine particles (particles less than 30 µm) are the most prevalent and dominate the water column. In water reuse systems (the focus of this book), fine particles (particles less than 30 µm) will dominate the water column. Figure 5.2 shows that 90% of the total particle mass in RAS systems is from individual particles of less than 30 µm (Chen et al. 1993b). This is why it is impractical and almost useless to use 60 µm drum filter screens to remove fine particles. Of course, large particles eventually become small particles, so rapid and efficient removal of the settleable (large >100 µm) particles is important, as it will minimize generation of the fine, more difficult to remove particles in the system. There have been several studies published on particle size distribution (Hannan, 1978; Easter, 1992; Chen et al. 1993b;; Cripps, 1992; Johnson and Chen, 2006).

Sedimentation techniques will not remove the fine particles from the water. This is because fine particles (<30 µm) have low settling velocities that make gravitational removal methods impractical. For example, fine particles need a retention time of several hours to settle a distance of 0.5 m. Sedimentation tanks are often regarded as being inefficient, but this opinion is usually caused by the settlement characteristics of fine particles, which require a lengthy retention time to settle (Chesness et al. 1975; Chiang and Lee, 1986), or because the sedimentation tank(s) were simply poorly designed to begin with (Henderson and Bromage, 1988).

5.5 Removal Mechanisms

There are three primary methods that are used to remove suspended solids from fish culture waters. These are:

- gravity separation
- filtration
- flotation

These classifications of methodology are based on the removal mechanisms used to effect the removal (flotation is sometimes considered as another kind of gravity separation, but it is a different principle of application so it is described separately). Large particles (larger than 100 µm) can be effectively removed by settling basins or mechanical screen filtration. However, fine particles cannot be removed effectively by either gravity separation or granular filtration methods. Granular filters are effective only in the removal of particles larger than 20 µm (Task Committee, 1986).

Gravity Separation. Gravity separation works on the principle of sedimentation and settling velocities. Unit processes in this category include clarifiers (settling tanks), tube settlers, and hydrocyclones. The actual process of particle removal can be accomplished with either screen, granular media (GM)**Error! Bookmark not defined.**, or porous media (PM) filters.

Filtration Removal. Particle removal from the water can be accomplished by one or more processes that contribute to the retention of particles in a filter. These are sedimentation, straining, Brownian diffusion, and interception. These processes are implemented in filtration systems by screen, granular media (GM), or porous media (PM) filters. In RAS applications, diffusion and interception have limited practical importance and will not be discussed (except as applied to foam fractionation, which is discussed later in this chapter).

Flotation Process. In a flotation process, particles attach onto air bubbles and are separated from water. The flotation process involves all the transport mechanisms that occur in a filtration process with the exception of straining.

The filtration techniques of sedimentation and straining are the most practical and commonly used methods, and are emphasized here. More detail on these methods is provided in the following sections.

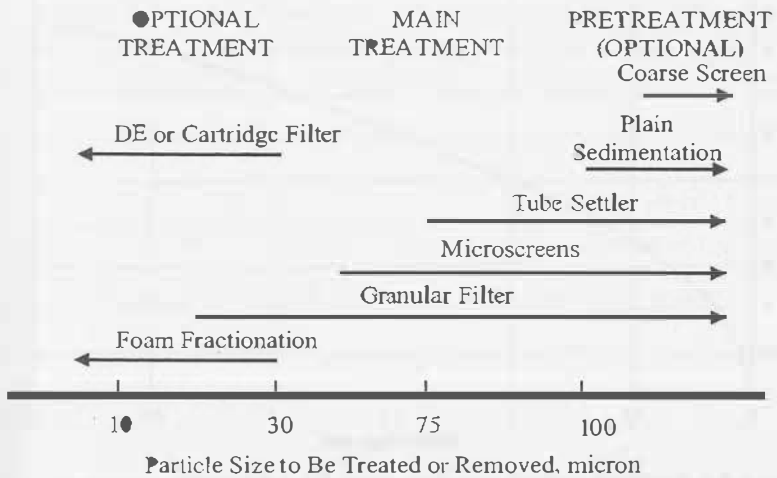


Figure 5.3 Solids removal processes and the particle size range in micron over which the processes are most effective (Timmons and Losordo, 1994).

Table 5.1 provides a summary of suspended solids removal techniques, their efficiency of removal and allowable hydraulic loading levels (Timmons and Losordo, 1994). Figure 5.3 categorizes technique appropriateness as related to TSS size characteristics.

When selecting a control process for TSS in a RAS, the following criteria should apply:

- hydraulic loading rate
- fine solids removal capability
- head loss
- water loss during filter backwash, and
- resistance to biofouling.

Table 5.1 Characterizations of Solids Removal Techniques for Recirculating Aquaculture Systems (Chen & Malone, 1991).

Technique	Solids Size Removed (micron)	Head Loss (m)	Hydraulic Loading m^3/d per m^2	TSS Removed (%)	Reference
Sedimentation	>100		24–94	40–60	EPA 1975
Settling tank			24–61		Liao 1980
Tube Settler			30-90		Muir 1978
Granular Media	>20	0.1–3	175-430	20–60	Muir, 1982; EPA 1975
Rapid sand filter			94–351	67–91	EPA 1975
			285	70–90	Mayo 1976
Pressure sand filters		2–20	115–700	50–95	Muir 1982; EPA 1975
Floating bead filter		0.8–6	1935		Wimberly 1990
Screen	>75	Negligi-ble	100–2,200		
Porous Media	>0.1		40–130		Muir 1982
Dissolved Earth	>0.1		29–59	>90	Muir 1982; EPA 1975
Cartridge	1–10	~ 5	1–10 gpm		
Hydrocyclones	1–75	14–35			Huguenin & Colt 1989; Wheaton 1977
Foam Fractionation	<30		290–280		Chen 1991
Ozonation	<30		see Chpt 11		

SEDIMENTATION

Sedimentation occurs due to the density difference between the solid particles and water. Assuming a particle to be heavier than water, under the force of gravity it will fall through the water with increasing speed until it reaches a terminal value for its settling velocity. Each discrete particle has an equilibrium settling velocity. Assuming spherical particles, the settling velocity can be calculated by the following equation (Montgomery, 1985):

$$V_s = \sqrt{\frac{4g(\rho_p - \rho)D_p}{3C_D\rho}} \tag{5.6}$$

For a small particle having a low Reynolds number (less than one, see Chapter 12), Stokes' Law applies, and Eq. 5.6 can be rewritten as

$$V_s = \frac{g(\rho_p - \rho)D_p^2}{18\mu} \tag{5.7}$$

Both Eqs. 5.6 and 5.7 indicate that denser and larger particles will settle out of water faster than smaller, less dense particles. This is true for all types of removal processes and why you should do everything possible to maintain large particle sizes. The best technique for maintaining large particle sizes is to remove the particles as quickly as possible from the fish culture vessel and before any pumping has occurred. Also, you should try to minimize any turbulence/falling water situations prior to the primary TSS capture event.

Reported Settling Velocities

Settling velocities for fish feces have been reported at 1.7–4.3 cm/s (Warren-Hansen, 1982). In tests we conducted, the authors measured settling velocities by using cylindrical beads (each 3 mm in length by 3 mm diameter) with a specific gravity of 1.05, to simulate tilapia feces. The beads exhibited settling velocities of 3.8 cm/s. These results are similar to the settling velocity reported for fecal matter by Warren-Hansen. Certain species produce fecal matter with even lower specific gravities (Robertson, 1992); trout fecal matter is can have a specific gravity as low as 1.005, in some instances. This is reflected in slower settling velocities of these wastes. Wong and Piedrahita (2000) reported that the median settling velocity on a mass-basis for the settleable solids from rainbow trout is 1.7 cm/s.

These reported velocities are consistent with the results obtained using Eq. 5.6 and as shown in Fig. 5.4. This similarity in settling velocity also suggests that the specific weight of feces would be similar. The authors measured the settling velocity of feed pellets, and found them to have a much more rapid settling velocity, e.g., 14 cm/s, than the plastic beads. This finding is similar to that reported by Juell (1991) of 15–33 cm/s. However, finer and/or less dense particles can be produced in RAS, which may settle at only 0.01 cm/s (IDEQ, 1998). These particles would not effectively settle in a settling basin or concentrate at the bottom center of dual-drain tanks, thus staying in the water column until removed by some other process.

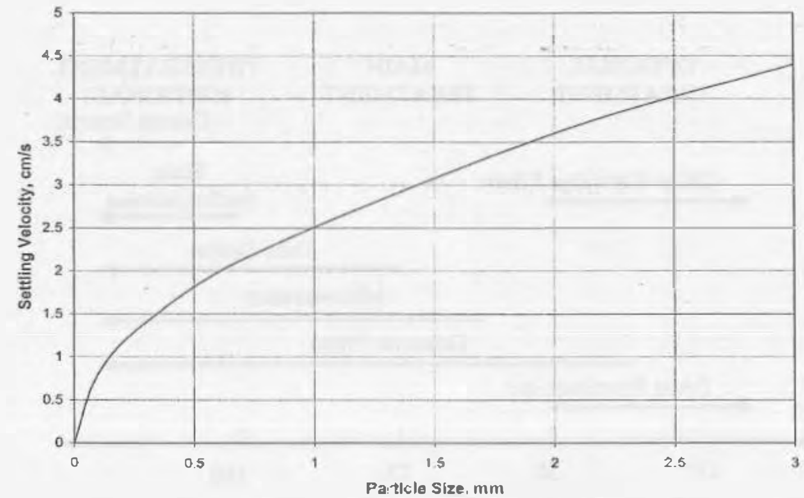


Figure 5.4 Predicted settling velocities using Eq. 5.6 (specific gravity of particle of 1.05).

Settling Basins

Settling basins are very effective if properly configured and operated. Sedimentation, i.e., gravity separation, is one of the simplest of technologies available to control particulate solids in process water and wastewater. Sedimentation basins require little energy input, are relatively inexpensive to install and operate, require no specialized operational skills, and can be easily incorporated into both new and existing facilities.

The disadvantages of sedimentation basins are:

- low hydraulic loading rates
- poor removal efficiency of small suspended solids (<100 μm)
- more labor intensive than mechanical filters
- additional floor space required compared to microscreen filters or granular media filters, and
- settled manure remains in the system until the settling basin is cleaned.

Innovative uses of vertical space over a settling bed or placing the settling bed in less expensive space can mitigate the problem of additional floor space. The cost disadvantage of added labor costs for cleaning settling basins will depend upon local labor rates. Areas with low cost labor would be more appropriate than areas with high cost

labor. The degree of solids dissolution, nutrient loading, and the re-suspension of solids that have settled and collected on the bottom of settling basins will be dependent upon how often the settling basin is cleaned, which then affects labor costs. Cripps and Kelly (1996) provide data relative to dissolution impacts. Henderson and Bromage (1988) estimated that settling ponds could capture an estimated 97% of their solids loading if resuspension of settled solids was not a factor. They suggest that settling basins are not effective in removing TSS when inlet concentrations are <10 mg/L or attaining effluent concentrations of <6 mg/L. Eliminating resuspension of TSS and dissolution of BOD and nutrients is difficult at best in most settling basins. Thus, settling basins will generally require further TSS treatment to meet the stringent removal criteria necessary to achieve mandated levels of TSS.

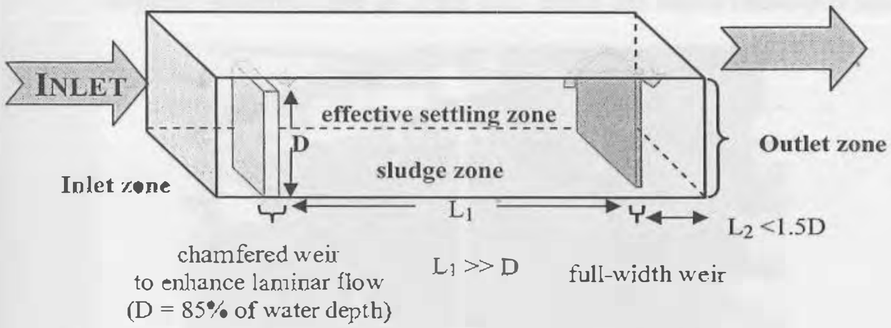


Figure 5.5 Four principal zones of a rectangular continuous flow sedimentation basin.

All continuous flow settling basins are conceptually divided into four zones according to function, Fig. 5.5. The inlet zone serves to uniformly distribute the suspension over the entire cross-section of the basin. Sedimentation occurs in the settling zone and, upon removal from the water column; the solids accumulate in the sludge zone. The clarified liquid is generally collected over the entire cross-section of the basin at the outlet zone and is discharged. Under ideal conditions (no mixing or turbulence), required retention time is the time required for a particle that starts at the top of the inlet zone and settles to the floor of the basin at or before the junction of the outlet zone. The key parameter for the design of settling basins is the volumetric flow of water per unit surface area of the basin or overflow rate (V_o):

$$V_o = Q/A_{sz} \tag{5.8}$$

"Rule of Thumb"

If you see any turbulence or currents in the settling basin, solids removal effectiveness will be reduced.

Any particle with a settling velocity (V_s) greater than the overflow rate (V_o) will settle out of suspension. Other particles, for which $V_s < V_o$, will be removed in the ratio V_s/V_o, depending upon their vertical position in the tank at the inlet (Hazen, 1904).

If one has access to TSS from a currently operating system similar to the one you intend to implement, then you can obtain a sample and test it to determine actual V_s for that particular waste; this is the ideal approach and has been described (Al-Layla et al. 1980). In doing these tests, use a settling column having an inside diameter greater than 13 cm. Results obtained from using narrower columns are not reliable, due to boundary layer resistance introduced by column walls. Use a test column that is the same depth as full-scale settling basin in the desired application. Without access to similar TSS, you can predict V_s using Eq. 5.6 or Fig. 5.4.

Settling basin designs were originally developed for municipal waste water treatment. Aquaculture waste is different, which is the reason for the erratic performance of settling basins when measured against the traditional approach of hydraulic retention time (τ) in the settling basin:

$$\tau = \frac{V_{basin}}{Q} \tag{5.9}$$

Hydraulic retention times of 15 to 30 minutes have been recommended for basin design (Liao and Mayo, 1974; Mudrak and Stark, 1981). Basin geometry is often used to maintain good settling conditions with recommended basin length to width ratios reported from 4:1 to 8:1 (Arceivala, 1983). It is also recommended that settling basins be designed so as to contain a minimum of 1 m (3.28 ft) of water depth (Liao and Mayo, 1974).

The overflow approach developed by Stechey and Trudell (1990) is an improvement over the hydraulic retention time approach. The overflow method provides much more effective TSS removal on a consistent basis. Stechey and Trudell (1990) recommend an overflow rate (V_o) for the design of settling basins in intensive salmonid aquaculture to be between 40–80 m^3/m^2 per day (982–1964 gpd/ft^2). These overflow rates translate to particle settling rates (V_s) equaling

0.046-0.092 cm/s. Translating this into easy to understand language, for every 1.0 Lpm of water flowing through the settling basin, 0.025 m^2 of surface area are required for settling (1.0 gpm flow per square foot).

Mudrak (1981) reported on the performance of several off-line settling basins used in intensive trout culture operations. He found that when the design overflow rate was at approximately 60 m^3/m^2 per day, the removal of settleable solids was 90% or greater, typically above 95%, although TSS removal was about 10% less. Also, there was no notable improvement in removal efficiencies when the loading rate was further reduced by as much as a factor of three, i.e., the conclusion is that a significant portion of TSS fine solids will not be removed by the settling basin. One design fundamental that must be kept in mind is that if you can see water currents in your settling basin, it will not efficiently remove the TSS except for the larger particles, e.g., >500 μm. Even if you double the settling basin floor area, this will not compensate effectively for a poorly designed settling basin where turbulence and mixing are present that are caused by ineffective inlet and outlet weir design.

The loading rates suggested above are applicable to off-line settling basins (the most widely used application in RAS). The Idaho Waste Management Guidelines for Aquaculture Operations (IDEQ,1998) suggests overflow rates for three typical settling basins used in aquaculture, i.e., full-flow, quiescent zone, and off-line (see Table 5.2).

Table 5.2 Surface Loading Rates for Settling Basin Design (IDEQ,1998)

	Surface Loading Rate m^3/m^2 per hr (ft^3/ft^2 per s)	gpm per ft^2
Full-flow settling basin	14.3 (0.013)	5.9
Quiescent zone	34.0 (0.031)	13.9
Off-line settling basin	1.66 (0.00151)	0.7

The Freshwater Institute (Shepherdstown, WV) has successfully applied this approach to concentrating the effluent coming off of drum filters in gravity thickening tanks. They used three off-line settling basins to capture and store solids from the intermittent backwash of three drum filters (see Fig. 5.6). The solids-laden backwash flow is introduced intermittently into the top and center of each tank. At the top of each tank, the flow is introduced within a cylinder with an open bottom that is centered within the tank. The cylinder improves the hydraulics of the tank's radial flow by directing the water to first flow down (underneath

the cylinder and towards the cone of the tank) and then up as it travels radially towards the effluent collection launder about the top circumference of the tank. These thickening tanks have performed well, capturing 97% of the solids discharged from the microscreen filter backwash flows, Table 5.3.

However, the solids stored within the thickening tanks are degrading, as indicated by the >10-fold increase in total ammonia-nitrogen across the thickening tanks. The thickened manure, pulled from the cone's bottom on a monthly basis, contains about 10% total suspended solids and about 1000 mg/L of total ammonia nitrogen.

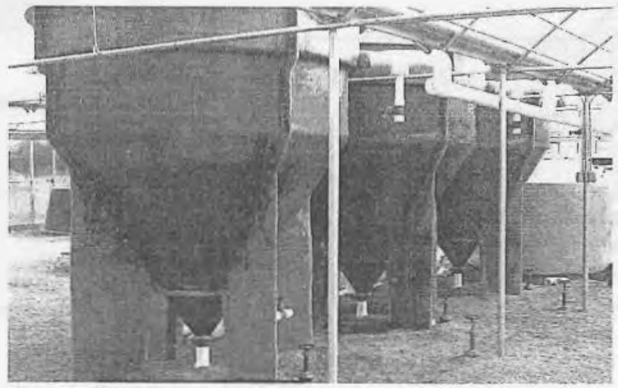


Figure 5.6 Off-line settling basins used for solids thickening and storage at the Freshwater Institute.

INLET AND OUTLETS FOR SETTLING BASINS

The transition of water to and from each of the zones must be accomplished so as to keep turbulence and mixing to an absolute minimum, as the rate of solids settlement is affected by the motion of the water.

The settling basin design most frequently used today in aquaculture places a single pipe at each end of a basin, thereby placing all three zones in the same vessel. This is the simplest but unfortunately also the worst design for inlet/outlet structures and settling basins. This type of point-source inflow and outflow produces extremely poor flow, which does not permit effective sedimentation. Short-circuiting is intensified as the

influent stream flows directly through the basin at relatively high velocity toward the discharge pipe. Consequently, a large portion of the fluid passes through the basin much quicker than the theoretical detention period or hydraulic retention time (τ) calculated based on the dimensions of the basin. To be effective, the design of a settling basin should address each of the zones as separate, identifiable functions of the basin. The design of the inlet and outlet structures is critical to the performance of the water being handled through the sedimentation process, and must address the specific function and constraints of each step within the process.

Table 5.3 Change in TSS, TAN, NO_2-N, and NO_3-N across Manure Thickening Tanks (Vinci et al. 2001)

	TSS mg/L	TAN mg/L	NO_2-N mg/L	NO_3-N mg/L
From the Drum Filter (Inflow to Thickening Tank)	5,147 ± 1,411	1.7 ± 0.1	0.19 ± 0.03	1.5 ± 0.1
Discharge from Thickening Tank	151 ± 24	19.5 ± 2.5	0.04 ± 0.01	0.7 ± 0.2

When designing inlet structures, the following factors must be considered (see IDEQ, 1988 for review):

- The influent stream should be introduced evenly across the entire cross-section of the settling zone.
- All flow through the settling zone should begin in even, horizontal path.
- The influent velocity to the settling zone should be slow enough
- to prevent excessive turbulence and mixing.

Inlets. Inlets should consist of a submerged inlet weir that separates the settling zone from the inlet zone. The inlet weir should extend across the full width of the settling basin, and should be submerged approximately 15% of the basin depth. The weir crest should be about 20 to 30 cm wide (8 to 12 inches) and have rounded edges to smooth the flow as it enters the settling zone. For circular clarifiers, the inlet is generally at the center of the basin. A baffle surrounding the inlet pipe serves to reduce turbulence and distribute the flow in a radial pattern through the full depth of the basin.

Outlets. Rectangular settling systems are more efficient than are circular settling tanks, but they require considerably more floor space. The sub functions of rectangular systems are more easily recognized as functional zones, Fig 5.5. These are the inlet zone, the settling zone, and the outlet zone. The outlet weir divides the settling zone from the outlet zone, as it skims clear water from the surface of the settling zone. The outlet weir should be designed and constructed so that it distributes the water exiting the settling zone at a uniform depth and velocity across its width. This is necessary to avoid generating currents and the accompanying turbulence in the settling zone. The outlet zone area should be the same width as the settling zone, and the length not less than 1.5 times the depth of the settling zone. For example, if the settling zone is 2.4 m (8 feet) wide, 9 m (30 feet) long, and 1.2 m (4 feet) deep, the outlet zone should be 2.4 m (8 feet) wide, 1.2 m (4 feet) deep, and at least 1.8 m (6 feet) long.

If a circular system is necessary due to space or other constraints, the design of the system must recognize and accommodate the same functions as are more easily accomplished in rectangular systems. There must be an inlet zone, a settlement zone, and an outlet zone. However, the boundaries between these zones are not clearly identifiable as they are in rectangular systems. These zones are created within the circular tank by the placement of inlets, the hydraulic movement of the water within the tank, the physical properties of the solids within the water, and the placement of the drain and outlet pipes.

In these cases, the flow towards the outlet is managed to create minimal turbulence particularly near the outlet pipe in the center of the tank. A circular weir can be installed that surround the outlet pipe, mostly to prevent floating solids from escaping the settling tank. Davidson and Summerfelt (2005) provide more information on the performance and design of circular 'radial flow' settlers within recirculating aquaculture systems. Radial flow settlers and other types of particle traps are now widely used to rough out a portion of the TSS contained in the concentrated but relatively low-volume discharge from the bottom-center drain in dual-drain circular tank systems (Twarowska et al., 1997; Davidson and Summerfelt, 2005; Veerapen et al., 2005; Johnson and Chen, 2006).

It is critical that the weir edge be level to assure a uniform discharge rate across the entire weir length. The weir discharge rate (volume of water discharged per unit length of weir per unit time) governs the length of the outlet weir. For weirs that are long in relation to the flow, i.e., having a low weir rate, a saw-toothed or V-notch edge is necessary for uniform discharge along the weir length. Weir discharge rates should be

400 to 600 m^3/d per meter length of the outlet weir (22 to 33 gpm/ft). More information on outlet weirs is provided in the literature (ASCE, 1959; EPA, 1975; Mudrak, 1981). Weir length should be maximized to the extent possible.

The total length of the settling basin is comprised of the actual settling zone plus the length (area) required for both the inlet and outlet zones. This total area requirement is often ignored and thus the settling basin will not perform as intended. In addition, remember that uncontrolled turbulence, i.e., mixing and stirring of the inlet waters with the incumbent waters will decrease the effectiveness of the settling process. The solution for this problem is to lengthen the sedimentation basin. Do not compromise on the size of the settling basin.

DESIGN EXAMPLE

Design a settling basin to remove 90% of the TSS particles greater than 0.5 mm in diameter with a design flow of 22.7 m^3/hr (100 gpm).

> **"RULE OF THUMB"**
> **Settling Basin Design**
> - basin floor area of 41 Lpm/m^2 (2,4 m^3/m^2/hr or 1 ft^2 per gpm of flow)
> - 250 to 410 Lpm per m width of weir for outflow (20 to 33 gpm per foot)
> - submerge inlet weir 15% of basin water depth
> - use 25 cm (10 inch) wide weirs and use rounded edges
> - maximize length of settling chamber as much as possible

<u>Solution</u>

Using 0.05cm/s as a design settling velocity (based upon IDEQ, 1998 recommendations for an off-line settling basin):

$$V_o = 0.05\ cm/s$$

Converting to flow per unit area

$$0.05\frac{cm}{s}\cdot 60\frac{s}{min}\cdot\frac{1\ m}{100\ cm}\cdot\frac{m^2}{m^2} = 0.030\frac{m^3}{m^2\ min}$$

$$(or\ 0.73\ gpm\ per\ ft^2)$$

Now determine the settling zone area, Equation 5.8:

$$A_{SZ} = \frac{Q}{V_o} = \frac{\left(22.7\frac{m^3}{hr}\right)}{\left(1.8\frac{m}{hr}\right)} = 12.6m^2(136 ft^2)$$

CHEMICAL AND POLYMER AIDS TO FLOCCULATION AND SETTLING

The Freshwater Institute recently completed research that determined optimum conditions for coagulating and flocculating waste biosolids from salmonid production systems using alum, ferric chloride, and/or commercially available polymers (Ebeling et al., 2003; 2004; Ebeling et al., 2006; Rishel and Ebeling, 2006). These studies determined optimum chemical and/or polymer dosages, mixing intervals and intensities, and settling times required for coagulating and flocculating waste biosolids within an off-line settling basin or belt filter. The coagulation-flocculation tests were carried out following the standard practice for coagulation-flocculation testing of water and wastewater. Based on these tests, recommendations were made on the design and operation of large-scale treatment systems using polymers, which included estimates for their treatment efficiency and performance.

In summary, settling basins can be used very effectively and economically for thickening settleable solids carried in backwash flows, at least if the basins are properly designed, configured, and operated. The key factors to having a successful settling basin are given in the rule of thumb box. Within recycle systems, however, use of settling basins is typically limited to relatively small radial-flow settlers and swirl separators, which are located to remove solids found in the concentrated and relatively low-volume flow exiting the bottom-center drain of a dual-drain culture tank (Davidson and Summerfelt, 2005; Veerapen et al., 2005; Johnson and Chen, 2006).

SETTLING DECK IN SETTLING CHAMBERS

A major objection to the use of settling basins is that they require a large floor area, and square footage of floor area can be expensive. If this is a problem, the "footprint" of the settling basin can be decreased by adding obstructions inside of the settling basin to increase rates of settling. Tube settlers, also known as "settling decks", can be used to do this. The basic function of the settling deck (tube settlers) is shown in Fig. 5.7 where the incoming flow is brought into the settling basin under the settling deck and forced to upflow to exit the chamber. In the process, solids settle within the tubes.

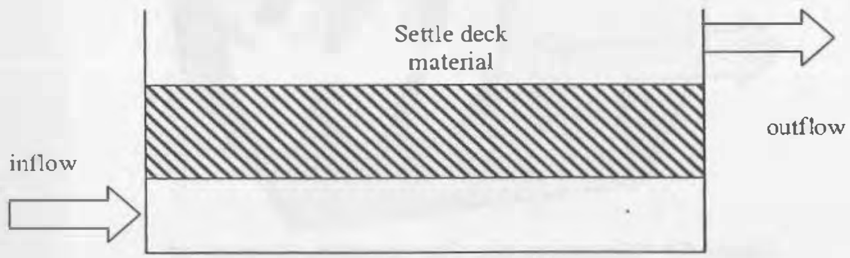


Figure 5.7 Settling chamber with settle deck to enhance solids removal efficiency.

Separation distance between the inclined surfaces is typically 5 cm (2 inches) with a total inclined length of 0.9 to 1.8 m (3 to 6 ft) (Gregory and Zabel, 1990). Tube or plate plastic media are usually manufactured in structured bundles of tubes or stacks of parallel plates in a variety of opening shapes (square, rectangular, tubes, hexagonal, chevron). In operation, influent water flows into a tube or plate settler and then upward through the inclined tubes or plates as solids settle on the plastic surfaces. Generally the inclination of the tubes or plates is between 45°–60° above horizontal. This angle provides for the greatest degree of gravity self-cleaning of settled solids out of the media and into the basin bottom. A fairly broad range of hydraulic loading rates have been suggested, e.g., 1.5 m^3/m^2 per hr (McLaughlin, 1981), 7.4 m^3/m^2 per hr (Libey, 1993), and 6.7 m^3/m^2 per hr (Parker, 1981). The disadvantage to the use of tube settlers is that tube or plate settlers do not adequately self-clean, so they must be periodically cleaned by other means to prevent biofouling (Gregory and Zabel, 1990; Tchobanoglous and Burton, 1991).

The tube diameter determines the effectiveness of the capture and the allowable hydraulic loading rates (m^3/s per m^2 or gpm/ft^2). The physics of this technique are that a particle will settle and rest on the bottom of an individual tube prior to the transporting fluid emerging from the top of the tube. Thus, tube diameter and face hydraulic loading rate on the settling deck will determine the size of the settling deck area to treat the solids laden influent. The water velocity through the tube should be maintained in a laminar flow region to prevent turbulence and resuspending settled particles, i.e., Re <2,300 (see Chapter 12).

The effectiveness of tube settlers in removing fine solids can be impressive. Easter (1992) reported 80% efficiency in the removal of particles larger than 70 µm and 55% in the removal of all particles larger than 1.5 µm using a multi-tube clarifier. As mentioned above, the biggest disadvantage of the tube settling approach is that the tubes must be cleaned and this can be an extremely time consuming and unpleasant task. Larger tube diameters require less cleaning, but they also have a lower efficiency in capturing solids. If you are going to use tube settlers, use tube diameters of at least 4 to 5 cm (1.5 to 2 inches). Using smaller diameter tubes will make the cleaning task all but impossible. The authors used some 1.5 cm diameter tubes in a rainbow trout system. As expected, the tubes collected feces, but cleaning it required removing the settling deck from the settling chamber and using high pressure hoses to flush the solids from the decking. This was a dirty, disgusting task. (As a personal note: it was this particular experience that inspired Timmons to develop the Cornell dual-drain approach. Ebeling just had his student interns, Sara and Kata clean the filters.)

Once the tubes begin to fill with fine solids settling out of the water flow, the water velocity rates through the tubes will increase due to reduced tube cross sectional area. As this happens and resistance to flow increases, water begins to seek a least resistance approach and will eventually simply bypass the tubes, thereby eliminating any solids capture at all. Therefore, periodic cleaning is necessary, but cleaning the settling deck is a dirty, nasty job that nobody likes to do and as a result is often neglected. In turn, neglect leads to poor performance of the settling device and subsequent deterioration in water quality. For this reason, we do not recommend tube settlers for use in highly loaded systems. But for lightly loaded larval or fry systems, they do work well.

MICROSCREEN FILTERS

Microscreen filters for filtration are popular because they require minimal labor and floor space in comparison to settling basins; they also backwash and remove the captured particle from the process flow before

substantial particle dissolution or BOD/nutrient leaching can occur. As in sedimentation processes, the head loss of a screen filter is small. Screen filters remove solids by virtue of physical restrictions (or straining) on a media when the mesh size of the screen is smaller than the particles in the wastewater. Microscreen filters are commercially available in a variety of different configurations. Typical microscreen filters used in aquaculture are the drum filter, Fig. 5.8, disk filter, Fig. 5.9, and inclined belt filter, Fig. 5.10.

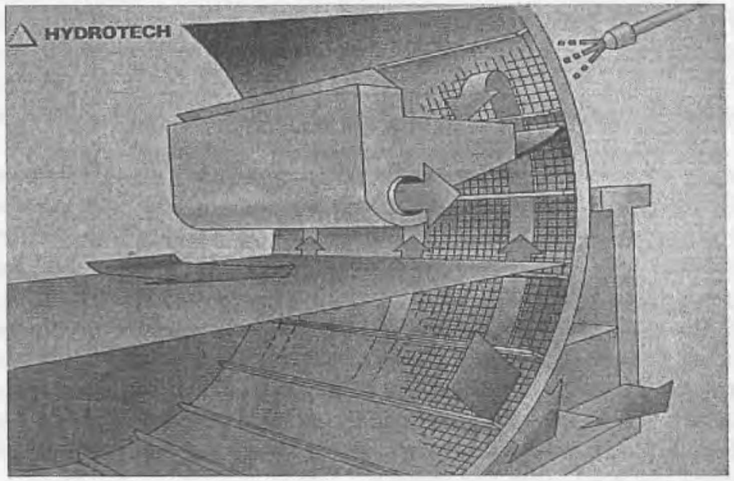


Figure 5.8 Microscreen drum filter operation. (Courtesy of Hydrotech AB, Vellinge Sweden & WMT, Inc., Baton Rouge, LA)

Vinci et al. (2001) summarized the relative operational advantages, drawbacks, and costs of three types of waste water filters (see Table 5.4). All three types of microscreen filters are similar in that they have a separate solids waste stream that must be managed to result in a complete waste management system (Chapter 6). This waste stream is a higher-solid, screen backwash flow. The backwash flow will vary in volume and solids content will vary based on several factors. These are the screen opening size, type of backwash control employed, frequency of backwash, and influent TSS load on the filter. Backwash flow is generally expressed as a percentage of the flow the filter treats, with reported backwash flows ranging from 0.2 to 1.5% of the treated flow (Summerfelt, 1999). This discharge is typically directed to an off-line settling pond (Brazil and Summerfelt, 2006), but has occasionally directed to a belt filter (Ebeling et al., 2006), created wetland

(Summerfelt et al., 1999), or other such device for final solids capture and storage.

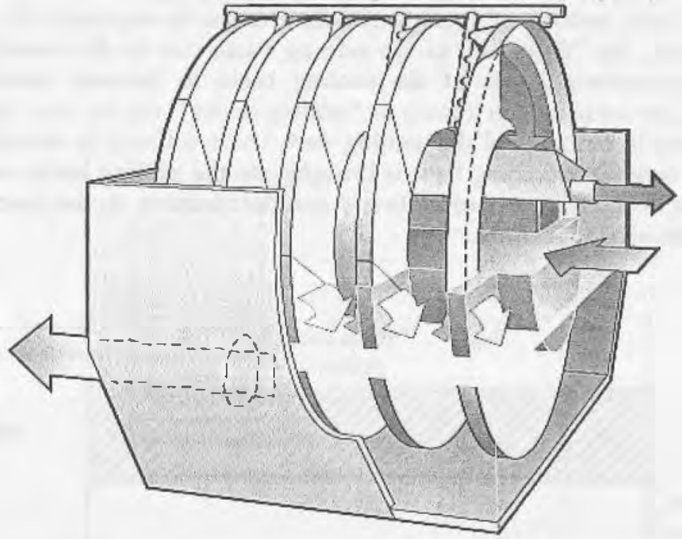


Figure 5.9 Microscreen disc filter operation. (Courtesy of Hydrotech AB, Vellinge Sweden & WMT, Inc., Baton Rouge, LA.)

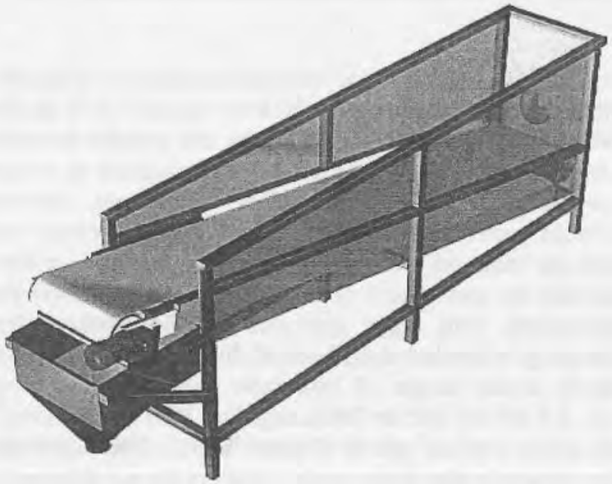


Figure 5.10 Inclined belt filter used for effluent solid removal. (Distributor: Sterner Aquatech AS; J. Ronhovde, personal communication.)

Table 5.4 Operational Advantages, and Drawbacks of 100 micron Mesh, 10 m^3/min Capacity Drum Filters, Disc Filters and Belt Filters (from Vinci et al. 2001)

Type of filter	Solid removal rate at 60 to 100 µm mesh, %		Main advantages	Any drawbacks
Drum filter	SS inlet <5 ng/L:	31-67	Intermittent backwash	
	SS inlet >50 mg/L:	68-94	Flushing reduced backwash volume	
Disc filter	SS inlet <5 mg/L:	25-68	Lowest investment costs	High backwash flow volume
	SS inlet > 50mg/L:	74-92	(without basic investments)	Grounding/crushing of bigger particles
Belt Filter	SS inlet <5 mg/L:	0-62	Gently removing particles	Relatively high investment costs at low flow (<3-5 m^3/)
	SS inlet >50 mg/L:	>89	Low maintenance work and costs Cost effective at high flow	

On a practical basis, simply assume that the screen size will define the smallest particles that will be removed by microscreening devices. However, to a limited degree, even particles smaller than the nominal screen mesh size can be trapped if several smaller particles bridge together and subsequently become trapped by the screen.

In addition to screen size, microscreen filter performance is dependent on the influent TSS concentration. Typical screen openings used in the treatment of aquacultural wastewater are 40–100 µm. In this screen size range, TSS removal can be between 30 and 80%.

Table 5.5 Example of Hydraulic Loadings, Screen Size, and Drum Filter Size as Part of an Overall Selection Process (Water Management Technologies, Baton Rouge LA, North America distributors of Hydrotech)

Flow capacity examples	Filter size	501	801	802	803	1201	1202	1203	1601	1602	1603	1604	1605	1606	1607	2005	2006	2007	2407	2408
	Filter opening (micron)	Maximum flow capacity (l/s)																		
Intake water from stream Lake or sea Max 10 mg/L SS	40	9	24	48	72	36	72	108	48	96	144	192	240	288	336	300	360	420	504	576
	60	11	30	60	90	45	90	135	60	120	180	240	300	360	420	375	450	525	630	720
	90	14	36	72	108	54	108	162	72	144	216	288	360	432	504	450	540	630	756	864
Recirculated fish farm Max 25 mg/L SS Cold Water	40	6	16	32	48	24	48	72	32	64	96	128	160	192	224	200	240	280	336	384
	60	8	22	44	66	33	66	99	44	88	132	176	220	264	308	275	330	385	462	528
	90	11	28	56	84	42	84	126	56	112	168	224	280	336	392	350	420	490	588	672
Recirculated fish farm Max 25 mg/L SS Warm Water	40	4	11	22	34	17	34	50	22	45	67	90	112	134	157	140	168	196	235	269
	60	6	15	31	46	23	46	69	31	62	92	123	154	185	216	193	231	270	323	370
	90	8	20	39	59	29	59	88	39	78	118	157	196	235	274	245	294	343	412	470
Outlet water from flow through type fish farm Max 15 mg/L SS	60	11	28	56	84	126	56	112	168	112	168	224	280	336	392	350	420	490	588	672
	90	13	34	68	102	153	68	136	204	136	204	272	340	408	476	425	510	595	714	816
	100	14	36	72	108	162	72	144	216	144	216	288	360	432	504	450	540	630	756	864
	500	20	52	104	130	234	104	208	312	208	312	416	520	624	728	650	780	910	1092	1248

Above figures shall be regarded as guidance. Please contact Hydrotech or representative for final sizing.
Figures are valid for standard H versions. Versions 2L and 2S may have a higher flow capacity in some applications.

Table 5.6 Performance Characteristics of a Disc Filter (Provided by Water Management Technologies, Baton Rouge, LA, North American distributors for Hydrotech products)

Flow capacity examples	Filter Size	1704/3	1704	1706/5	1706	1708/7	1708	2102/1	2102	2104/3	2104	2106/5	2106	2108/7	2108	2110/9	2110	2112/11	2112
	Filter opening (micron)	Maximum flow capacity (l/s)																	
Intake water from stream, lake or sea Max 10 mg/L SS	40	180	200	200	200	200	200	80	160	240	320	400	480	560	640	720	800	800	800
	60	180	200	200	200	200	200	80	160	240	320	400	480	560	640	720	800	800	800
	90	180	200	200	200	200	200	80	160	240	320	400	480	560	640	720	800	800	800
Recirculated Fish Farm Max 25 mg/l SS Cold Water	40	130	174	200	200	200	200	70	140	210	279	349	419	489	559	629	698	768	800
	60	179	200	200	200	200	200	80	160	240	320	400	480	560	640	720	800	800	800
	90	180	200	200	200	200	200	80	160	240	320	400	480	560	640	720	800	800	800
Recirculated Fish Farm Max 25 mg/L SS Warm Water	40	91	122	152	183	200	200	49	98	147	196	244	293	342	391	440	489	538	587
	60	125	167	200	200	200	200	67	134	202	269	336	403	471	538	605	672	739	800
	90	180	200	200	200	200	200	80	160	240	320	400	480	560	640	720	800	800	800
Outlet water from flow through type Fish Farm Max 15 mg/L SS	30	147	196	200	200	200	200	79	157	236	314	393	471	550	629	707	786	800	800
	40	179	200	200	200	200	200	80	160	240	320	400	480	560	640	720	800	800	800
	60	180	200	200	200	200	200	80	160	240	320	400	480	560	640	720	800	800	800
	90	180	200	200	200	200	200	80	160	240	320	400	480	560	640	720	800	800	800

Above figures shall be regarded as guidance. Please contact Hydrotech or representative for final sizing.

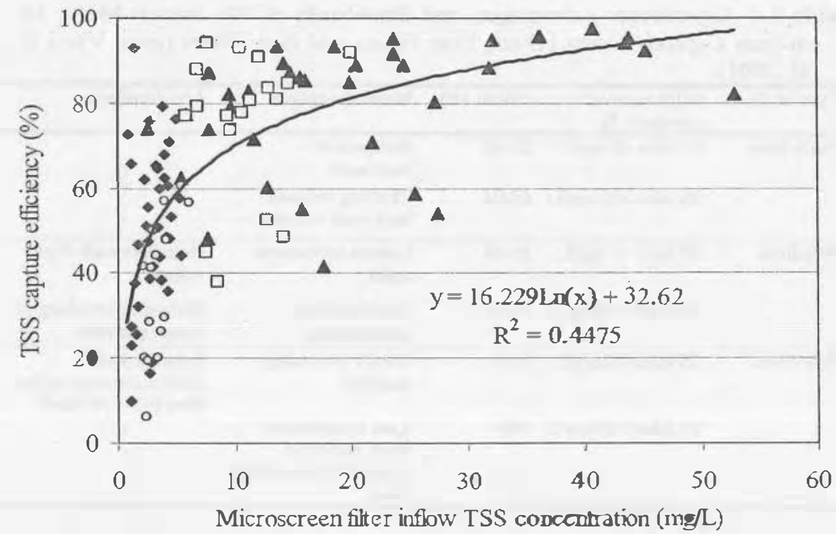


Figure 5.11 Removal efficiencies versus inlet TSS concentration (Summerfelt et al. 2001).

Summerfelt (2001) summarized a large amount of work that quantified the effectiveness of removing TSS using drum filters with screen sizes from 60 to 90 μm (see Fig. 5.11). This data was collected from four microscreen filters (60 to 90 μm) used at the Freshwater Institute (Shepherdstown, WV): one microscreen drum filter (○) treated the flow discharged from the elevated drains of dual-drain culture tanks in a water reuse system; another microscreen Triangle™ filter (△) sieved the bottom flow from a dual-drain culture tank in a single-pass system; another microscreen drum filter (□) treated the combined effluent from a reuse system and a single-pass system; and the other microscreen drum filter (●) sieved the effluent from a single-pass system.

The data shown in Fig. 5.11 shows a trend of increasing removal efficiency with increasing influent TSS concentration. In other words, the dirtier the water coming into the filter, the better the filter works.

Smaller filter pore sizes increase the removal rate of TSS to a certain extent. Based on particle size distribution analysis of hatchery effluent (Cripps, 1993), an increased removal rate of solids is expected when using increasingly smaller filter pore size below 200 μm. There are limits to this increasing removal rate, however. In a follow-up study by Cripps (1995), he found that screen sizes below 60–100 μm no longer improved solids removal. This efficiency limit was confirmed by Kelly et al.

(1997) who studied the particle removal in effluent water from two salmon hatcheries using test sieves with four mesh sizes between 30 and 200 μm, Fig. 5.12.

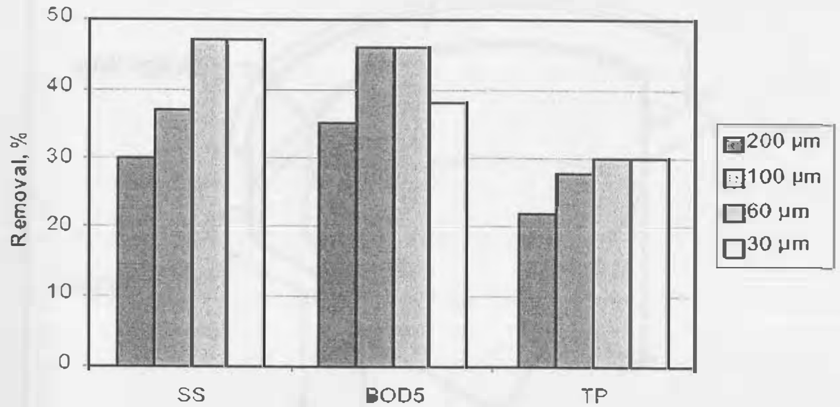
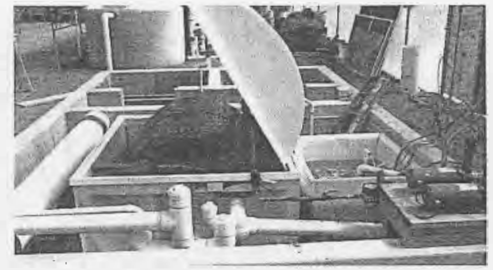


Figure 5.12 Daily average removal of Suspended Solids (SS), Biochemical Oxygen Demand (BOD_5) and Total Phosphorus (TP) after passage through four apertures (200, 100, 60 and 30 micron) (Kelly et al. 1997).

Typically, the removal rate was significantly higher during tank cleaning operations (SS: 10–100 mg/L) compared with the removal during routine flow (SS <6 mg/L) throughout the day. The 60 μm aperture was estimated to reduce the main daily BOD_5, SS and TP outflow concentrations by 45, 46 and 30%, respectively. Consequently, the authors concluded that to achieve maximum effectiveness of solids filtration of waste waters from tank farms, the filtration system should use a screen with an aperture of 60 to 100 μm. This conclusion is supported by returning to Fig. 5.2 which shows that 80 to 90% of the solid particles smaller than 130 μm are in fact less than 30 μm in size, and using a screen that small is impractical due to pressure losses and excessive backwashing requirements. In slight contrast, Wedekind et al. (1995) did find improvement in solids removal when the screen size was reduced from 60 to 30 μm in an intensive rainbow trout production system. However, a markedly increased volume of backwash water (a bad thing) and sludge (a good thing) resulted when reducing that screen pore size.

SIZING AND MANAGING DRUM FILTERS

The use of the Cornell dual-drain system in conjunction with drum filters can produce a 10-fold higher concentration of TSS in the influent water to the drum filter. This means that the hydraulic capacity of the drum filter will be reduced by approximately 30% when compared to non dual-drain influents (see Table 5.5 which shows hydraulic capacity vs. varying strengths of the intake TSS load). Consult your supplier carefully in this regard and be careful when comparing the drum filters of competing manufacturers so that the comparison is made using the same influent TSS concentrations. Table 5.5 shows the relationship between unit selection and hydraulic loading, screen size, and influent TSS concentrations. Filter screens require regular maintenance. If a microscreen filter is properly sized, the drum filter should not rotate and clean itself any more frequently than every 2 or 3 minutes; more frequent rotations indicates that the screen needs maintenance, or it has an overload of suspended solids (this means you needed a bigger unit!). Backwash flow (usually the discharge from facility) will be 0.2 to 2% of treated flow, depending largely on the frequency of backwash.



Increased backwashing frequency is a clear sign that screen maintenance is needed. In any case, pay careful attention to the manufacturer's recommendations for maintenance intervals and processes.

DISC FILTERS

Disc filters are an alternative to drum filters (see Fig. 5.9). Disc filters are less common in the USA, but are used in Europe especially on farms with high volumetric flows that require suspended solids (TSS) removal. Disc filters are generally less expensive than drum filters when treating high volumetric discharges, e.g., flows 8 m^3/min (>2,000 gpm). A criticism of disc filters is that because of the vertical orientation of the filter screens, TSS can be retained longer before removal. This longer retention time could cause TSS breakup into finer particles and perhaps additional BOD and nutrient leaching. Even if additional breakup of TSS occurs, the size range would still be much larger than 60 micron, which means practically the same TSS removal efficiencies should occur with a disc as a drum filter. A performance table is given for disc filters (see Table 5.6, courtesy of Water Management Technologies, Baton Rouge, LA, North American distributors for Hydrotech products).

SWIRL SEPARATORS

Swirl separators and tea-cup settlers take advantage of water circulation and currents to concentrate the solids in the center/bottom of a tank and improve separation. The Cornell dual-drain is effective because the tanks are round and are operated as swirl separators. The rotational flow of the inlet water imparts a slight centrifugal motion in the particles that causes the heavier particulate material to move to (or remain at) the outer portion of the vessel. Simultaneously, the particles are affected by gravity, which causes them to fall through the water, and move towards the bottom center drain due to a secondary current created by the rotation of the water. Here, at the bottom center drain, a small percentage of the total flow is removed, which is referred to as the underflow.

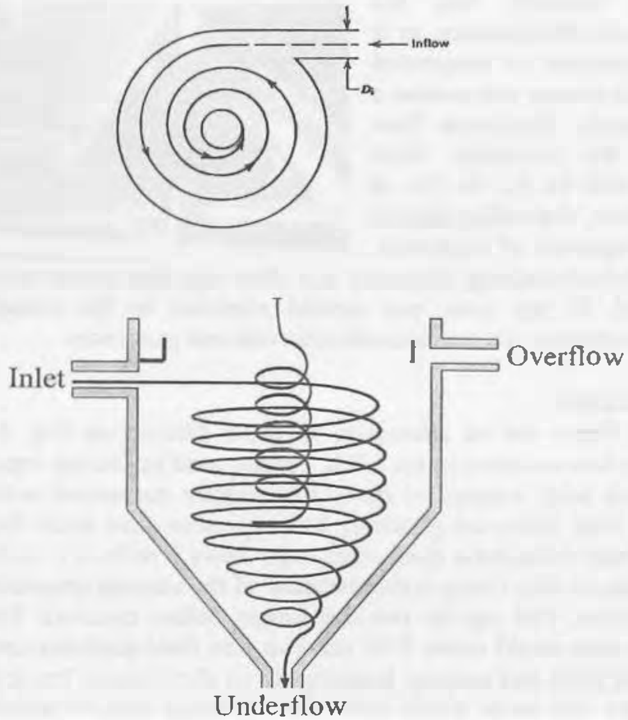


Figure 5.13a Design concepts of a swirl separator.

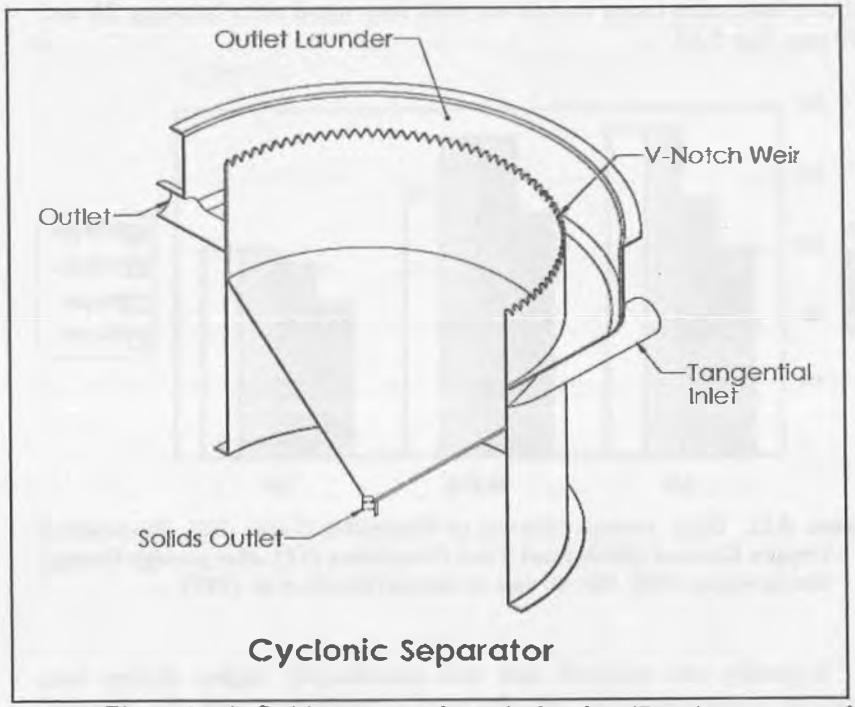


Figure 5.13b Swirl separator schematic drawing. (Drawing courtesy of HE Products, PO Box 145, Shandon OH 45063, 513-738-4333)

The inlet is typically placed about 1/3 diameter below the top of the water column with the flow directed tangentially with respect to the vessel walls. Figure 5.13a and b illustrates the design concepts of a swirl separator. Paul et al. (1991), Tchobanoglous and Burton (1991), and Veerapen et al. (2005) provide additional design approaches and theory for vortex driven settling basins.

Davidson et al. (2005) evaluated a radial flow separator, which is an adaptation of a conventional swirl separator design, see Figure 13c. They found the radial flow separator nearly twice as effective as a swirl separator at the same surface loading rates and same sized units. The authors consider that design parameters and constraints are similar between either unit application. Radial flow separators can provide better performance, mostly due to the low density of aquaculture type solids, which makes the center cylinder more critical to prevent light solids from escaping the unit prematurely.

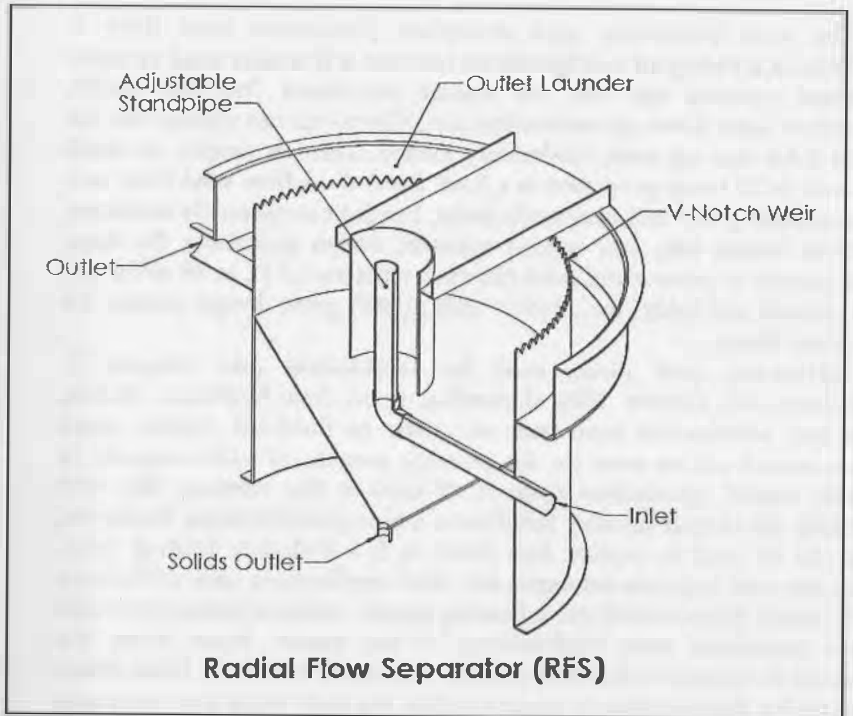


Figure 5.13c Design concepts of a radial flow separator. (Drawing courtesy of HE Products, PO Box 145, Shandon OH 45063, 513-738-4333)

Loading rates for swirl separators depend upon the size and specific gravity of the particles to be treated, which is similar to how loading rates are selected for settling basins. Although many different loading rates have been reported in the literature, the authors (Davidson and Summerfelt, 2005) have experience using a hydraulic loading rate that is approximately four times greater than the recommended loading rates for conventional off-line settling basins, or 10 m^3/m^2 per hr (4 gpm per ft^2) can provide effective treatment. This means that the footprint area requirements for handling the same amount of water have been reduced by a factor of four. Some engineers will use even higher loading rates, but hydraulic retention times should be maintained at a minimum of 30 seconds. If loading rates are increased, then the volume of the vessel must be correspondingly increased so that the minimum 30 second

hydraulic retention rate is preserved. Note also that some swirl separators are operated without a continuous underflow, such that captured biosolids are stored in the base of the swirl separator until they are manually flushed, approximately 1-2 times daily.

"Rule of Thumb"

Swirl or Radial Flow Separators

- hydraulic loading of 122 to 204 Lpm/m^2 (3 to 5 gpm per ft^2)
- underflow of 5 to 15% of total flow
- tangential inlet flow or use of central cylinder
- inlet submerged 1/3 vessel diameter or with central cylinder
- best used with dual-drain tanks

Swirl separators are not inexpensive, and just as settling basins and drum filters, they are not effective at removing fine solids (diameter <50 μm). However, they can be quite effective in removing TSS: Davidson and Summerfelt (2005) report nearly 40% removal efficiency. However, in the same study, Davidson and Summerfelt (2005) determined that a radial-flow settler operated under the same hydraulic loading rate in the same system would remove almost 80% of the TSS, which doubled the removal efficiency produced by a swirl separator.

One approach has been to use a commercially available swirl separator developed by SINTEF NHL (Trondheim, Norway) called the Eco-Trap™. Twarowska et al. (1997) reported that the Eco-Trap™ swirl separator that took 5% of the total center drain flow (the underflow with concentrated solids) removed 80% ±16% of the solids. The surface loading rate on this unit, which accepted 16 L/min of flow, was approximately 5.6 m/h[a] (2.3 gpm/ft^2). Referring back to Table 5.2, this loading rate is less than a full-flow settling basin (14.3 m/h) but more than an off-line settling basin (1.66 m/h). The swirl settling feature has reduced the floor area requirement of the settling process by a factor of 3.4, which is consistent with the earlier note that surface loadings could be increased by a factor of four compared to conventional settling basins.

[a] Note that m^3/m^2 per hour is equivalent to m/h or meter per hour.

GRANULAR MEDIA (GM) FILTERS

Granular media filtration involves passage of water through a bed of granular material (media) and deposition of solids onto the media. This type of filtration system is generally classified as packed-bed or depth filtration (Crites and Tchobanoglous, 1998). The major mechanisms that function to remove the particulates in a packed-bed filter are straining, sedimentation, impaction, interception, adhesion, flocculation, chemical adsorption, physical adsorption, and biological growth (Tchobanoglous and Burton, 1991). However, straining has been identified as the principal mechanism for the removal of suspended solids in the filtration of secondary effluent from biological treatment processes (Tchobanoglous and Eliassen, 1970).

Granular-medium filters are often used in combination with other treatment processes to increase the level of treatment. These filters provide increased removal of solids, phosphorus, algae, turbidity, and pathogens. In the case of phosphorus removal, the water is pre-treated by adding chemicals that precipitate phosphorus, i.e. alum or ferric chloride (Ebeling et al., 2003; 2004; Rishel and Ebeling, 2006). Precipitated phosphorus is then subsequently removed in the particulate form by the granular-medium filter. It is important to note that the backwash from these filters will have high total phosphorus levels.

There are several types of granular-medium filters. These are: conventional monomedium downflow filter, conventional dual-medium downflow filter, conventional monomedium deep-bed downflow filter, continuous backwash deep-bed upflow filter, pulsed-bed filter, traveling-bridge filter, synthetic-medium filter, and pressure filter. The basis of operation of all these filter types is the same; they all use the previously stated removal mechanisms, and they all require a backwash or cleaning process to regenerate the particulate removal capacity of the filter (Crites and Tchobanoglous, 1998). For example, in the case of the continuous backwash deep-bed upflow filter, process water is evenly introduced at the filter bottom where it upflows through a sand bed. The filtered water flows out of the sand bed, into an overflow weir, and is discharged from the filter. Concurrent to this filtration, sand and solids trapped by the sand are drawn downward within the sand bed into the suction of an airlift chamber in the center of the filter. The airlift carries the sand and solids to the top of the filter into a reject compartment. Because the discharge weir from the reject compartment is lower than the filtrate weir, the reject compartment continuously receives a small flow of filtered water that carries solids out of the filter. This backwash flow also acts to clean sand that is settling back down to the top of the sand bed after having been airlifted to the reject chamber.

The most commonly used downflow pressurized sand filter is available in a variety of configurations because it is widely used in water treatment systems and can be readily purchased "off the shelf". Downflow sand filters (or swimming pool filters) are not appropriate for use in RAS that are even moderately loaded. There is simply so much TSS and BOD being generated in a RAS that a downflow sand filter will be constantly going into backwash mode. For the exceptionally stubborn, or those having very low loaded systems, design guidelines for large scale gravity or pressurized sand filtration units are of 12 to 30 m^3/hr per m^2 (Metcalf and Eddy, Inc., 1991). Bell (1962) gives design criteria for diatomite filters.

Upflowing sand filters used for biofiltration (see Chapter 7: Biofilters) will capture TSS, depending upon their hydraulic loading rates and whether the sand beds are static or fluidized. Upflow sand filters should not be used for the primary purpose of solids capture in heavily loaded aquaculture systems. If used in this manner, they will probably fail in their primary function of a biological filtration. However, they can be used to capture fine solids at low hydraulic loading rates, when the sand beds are not expanded. RAS applications have effectively used "bead" filters, which use a floating plastic media to reduce the water losses associated with backwashing of the media. Bead filters are operated to capture solids and provide biological filtration. Bead filters also require backwashing to remove solids, but their water loss compared to traditional downflow sand beds is very small. Figure 5.14 shows an industry supplied bead filter.

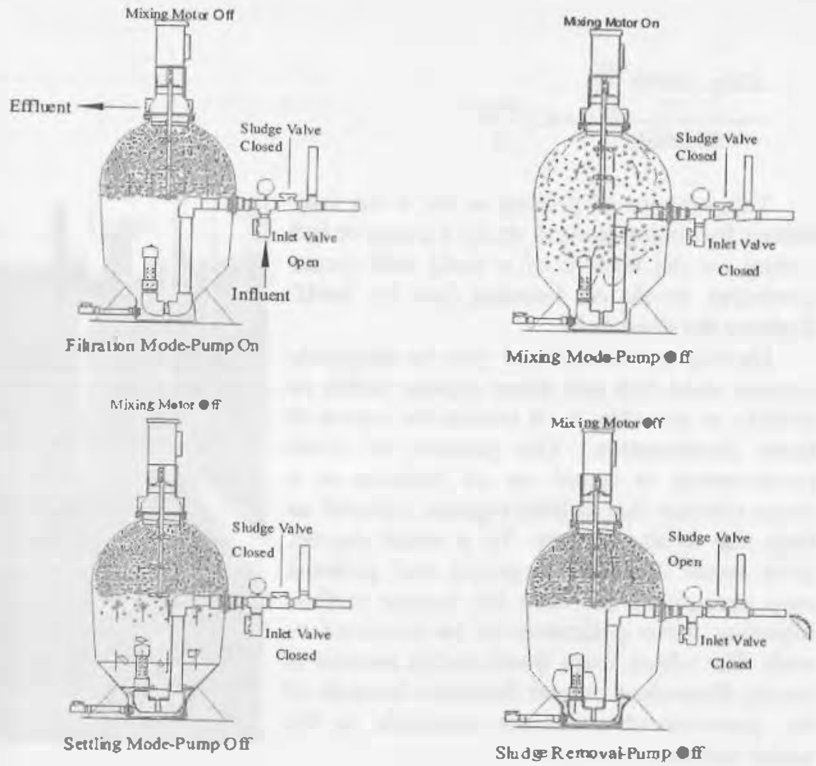


Figure 5.14 Bead filter operation. (Courtesy of Aquaculture Systems Technologies, LLC, New Orleans, LA)

Bead filters can be designed to obtain most of the benefits of static sand filters without incurring the high water losses during the backwashing operation. These filters are multi-functional, providing both solids removal and biofiltration processes (Golz et al. 1999; Malone and Beecher, 2000). Pressurized bead filters contain floating plastic beads that provide a medium to which nitrifying bacteria attach. The floating plastic beads commonly used in these filters are 3–5 mm (0.12-0.20 inch) diameter polyethylene spheres that have moderately high specific surface areas (1,145 m^2/m^3 (350 ft^2/ft^3) (Malone et al. 1993; Malone and Beecher, 2000). In operation, water is typically pumped into the bead filter through a media retaining screen located at the bottom of the bead chamber, Fig. 5.14. Particles are captured within the filter as the water flows upward through the floating bead bed. The bead bed floats against a screen installed at the top of the pressure vessel, which is used to retain

the beads while allowing the filtered water flow to pass out through the top of the filter. Operating pressures, typically 0.34–1.02 atm gauge pressure (5–15 psig), and increase as solids capture across the filter increases. To maintain adequate flow through the bead filter as solids build up, the filter is backwashed through the use of a motor-driven propeller mechanism or through injected bubbles. Backwashing occurs as the influent flow is stopped and the beads are turbulently disturbed, which breaks loose any collected solids. The solids are then removed from the filter through the media retaining screen located at the base of the filter, and normal flow is resumed (Malone and Beecher, 2000).

Solids captured by the filters are retained within the filter until the unit is backwashed. Typically, the plastic-bead filter unit is backwashed every 24 hours. During this retention period, 30-40% of the total retained solids will decay (Chen et al., 1993a). This is a very undesirable activity, because the entrapped solids begin to dissolve and mineralize, which adds soluble BOD and ammonia loads to the system. More frequent removal of these solids is the only way to minimize this condition. There is a complex relationship between plastic-bead filter backwash intensity and frequency, solids degradation, and nitrification efficiency (Golz et al. 1999). The fact that the pressurized bead filter collects solids so effectively means that heterotrophic bacteria populations can grow and become a problem if the filter is not backwashed frequently. Heterotrophic activity in the filter depletes oxygen and generates ammonia as protein contained in the captured solids is metabolized. While backwashing will remove solids and unwanted bacterial growth, too frequent backwashing will also remove beneficial nitrifying bacteria. Obtaining optimum performance from the bead filter requires careful management of the intensity and frequency of backwashing these filters (Golz et al. 1999).

Porous Media (PM) Filters

Porous media (PM) filters are often known as point-of-use devices and are used to treat low flow rates. They are very susceptible to clogging, even at low influent TSS concentrations, and experience high head losses. On a practical basis, the requirements for high recharging reconditioning or replacement of the media as the filters become plugged or restrictive to flow prohibit the use of porous filters as the principle solids capture device in an intensive RAS. However, PM filters do remove fine solids and are thus effective polishing units. PM filters can be used to treat incoming water to an RAS or as an ultra polishing step in a particular RAS application, e.g., incubation system. Two examples of PM filters are diatomaceous earth filters (DE) and

cartridge filters. Cartridge filters use disposable cartridges of different sizes. Like DE filters, cartridge filters remove particles by straining due to their thin media. Therefore, the removal of particles smaller than the pore size by cartridge filters is low. However, because of their fine pore size, both DE and cartridge filters can remove very small solid particles (<1 μm).

Foam Fractionation

Ugly foam in fish tanks is something we all detest seeing and something we all want to eliminate. However, as a practical bit of advice, if you can live with the foam, then don't try to get rid of it. Foam removal is typically a messy, unpredictable, and erratic process. But if you are convinced that you must remove the foam, this section will give you the basics of how to go about it. Remember that removal of wastes by foam fractionation is no substitute for effective primary TSS removal. Let's also include in this primary removal category the quick removal of dead fish, which consist of a high percentage of protein (assume 70% dry matter basis). Foaming is the result of the presence of surfactants, and surfactants are protein. Thus, a dead fish that is ground up through a pump or is left in the tank to decompose will provide a large amount of surfactant, which will in turn result in immediate and noticeable foaming.

Practical Example: Foaming

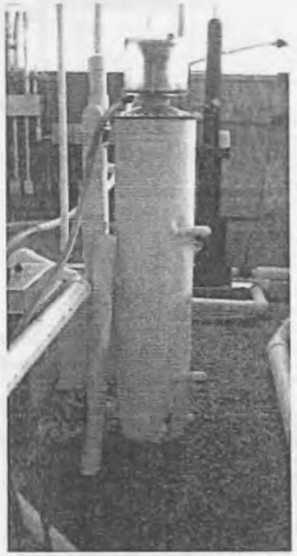
If you ever see an unexpected level of foaming in your system, start looking for a decomposing fish. A simple example will illustrate the point. Assume that in a tank of 10,000 fish, that you will lose 5% of the animals over a 200 day growth period. This means you are losing an average of 2.5 fish each day. Using a carrying capacity of 120 kg/m^3, 2.5 fish at 1 kg each (20% dry matter) would be the equivalent of:

$$10{,}000\,fish \cdot \frac{1kg}{fish} \cdot \frac{m^3}{120kg} = 8.83m^3 = 83{,}000L$$

$$2.5\,fish \cdot \frac{1000g_{flesh}}{fish} \cdot \frac{0.2g_{DM}}{1.0g_{flesh}} \cdot \frac{0.7g_{protein}}{1.0g_{DM}} = 350g_{protein}$$

Now, the concentration of this amount of pure protein (a foaming agent) in the 83,000 L of water is:

$$\frac{350g \cdot 1000\frac{mg}{g}}{83{,}000L} = 4.2\frac{mg}{L}$$



This amount of protein in the water (say from a fish being ground up by a pump or left rotting on the bottom of a tank) will create enormous levels of foaming just by itself. Remove the dead fish!

Having now sensitized you to diligently remove dead fish and other organic solids as quickly as possible, we'll review the basics of foam fractionation. The process of foam fractionation is based on air bubbles in a water column that collect organic material as they rise to the surface. To a small degree, even some dissolved organics and proteins may precipitate out onto the bubble surface allowing these pollutants to be removed as well. The whole foam fractionation process is totally dependent on and functions because of the presence of surfactant materials in the water column.

Surfactant molecules are polar with a hydrophilic (positively (cationic) or negatively (anionic) charged or nonionic) and a hydrophobic end. The hydrophobic end thus "pokes" itself into the air bubble leaving the hydrophilic end in the water. If the hydrophilic end is negatively charged and is now sticking out into the water column then it attracts positively charged materials in the water column. These positively charged materials in turn attract negatively charged particles that "stick" to them. Figure 5.15 illustrates the properties of surfactant molecules attached to an air bubble. Charged surfaces of an air bubble attract both negative and positive fine solids and bacteria.

The surfactant materials and other collected particles attached to the air bubbles will rise to the top of the water column as foam. The foam can then be removed from the water column. Foam fractionation is considered one of the few processes that are effective in removal of fine solids from a RAS. In fact, foam condensate consists mostly of dissolved organics and particles smaller than 30 μm. Foam fractionation is more effective in seawater systems than in freshwater systems, since the surface tension in seawater is higher than in fresh water. Thus, foam

fractionation is typically an integral component of recirculating seawater systems.

Removing and disposing of the foam is one of the problems encountered when using foam fractionators (Malone and Beecher, 2000). Plan ahead to determine what you will do with the foam condensate, e.g., collection vats that must be emptied or direct drainage to a floor drain. Foam fractionators have been well reviewed in the literature (Chen, 1991; Chen et al. 1992; Chen et al. 1994a, b; Lemlich, 1972). Typical foam fractionators are depicted in Fig. 5.16 (A = airlift type and B = venturi type).

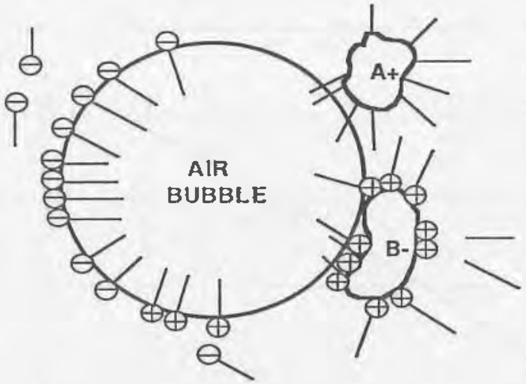


Figure 5.15 Surfactant molecule properties.

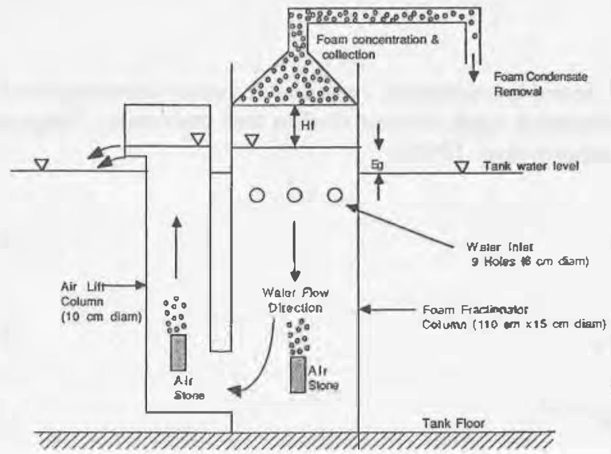


Figure 5.16A Airlift type foam fractionator.

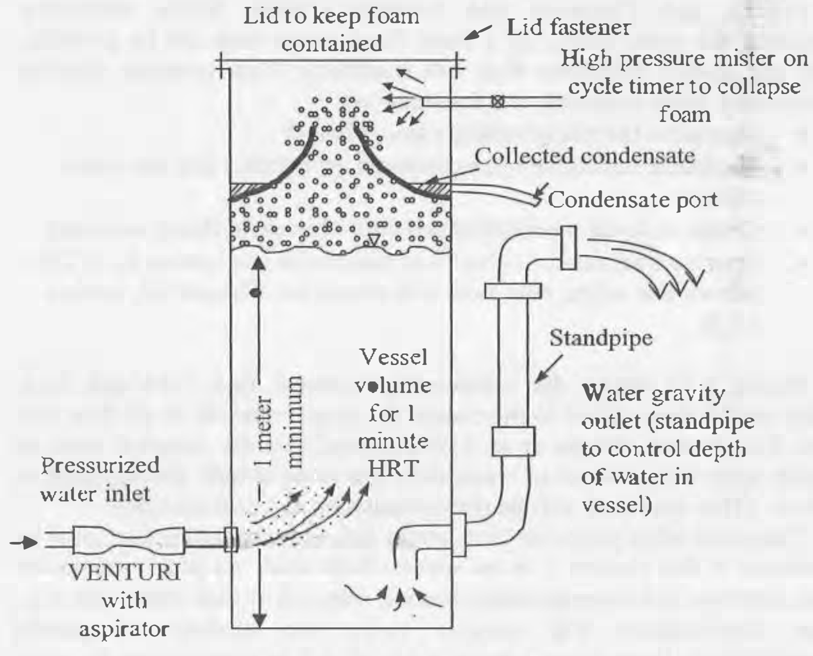


Figure 5.16B Venturi type foam fractionator.

The use of foam fractionators requires a basic knowledge of the fundamental parameters used in their design and operation. They are as follows (see Timmons et al. 1995):

$$U_g = \frac{Q_{air}}{A} \tag{5.10}$$

$$E_g = 4.1\,U_g^{0.83} \tag{5.11}$$

$$U_b = 0.21e^{-25.8U_g} \tag{5.12}$$

Where U_g is the superficial gas velocity (m^3/m^2/sec), E_g is the gas holdup (fraction), and U_b is the bubble rising velocity (m/s).

Predicting foam fractionator performance is problematic at best. For those interested, see Chen et al. (1992), Timmons et al. (1995), Weeks et al. (1992), and Timmons and Losordo (1994). While absolutely predicting the performance of a foam fractionator may not be possible, there are certain principles that will maximize foam creation, thereby maximizing waste removal, in a fractionator:

- Maximize the rising bubble's travel length
- Maximize the contact time between air bubbles and the water column
- Create as small a bubble as possible (decreases rising velocity)
- Operate fractionator columns at maximum gas holdup E_g of 25% (above this value, slug flow will occur; see Chapter 12, section 12.5)

Figure 5.17 shows the relationship between Eqs. 5.10 and 5.11, which must be calculated to determine the proper amount of air flow in a foam fractionator. Weeks et al. (1992) found that the removal rates of protein were independent of water flow but were simply proportional to airflow. (This approach will be demonstrated in the next section).

There are other pertinent facts about foam fractionation that must be addressed if this system is to be successfully used. As pH is increased, foam creation and corresponding protein removal is also increased, e.g., foam fractionators will remove twice the amount of protein concentrations from water having a pH of 8.3 in comparison to water with a pH of 5.3. And while use of smaller bubbles improves the overall

fractionation process, generally gas bubbles will be between 2 and 3 mm as created by a wide variety of typical scintered glass air stones. It is difficult to create smaller bubbles. The authors have achieved some success in creating smaller bubbles by using venturis when a centrifugal pump is used to bring water to the foam fractionator. However, if you are using the type of fractionator that is based upon using air lift pumps (see Fig. 5.16A), using a venturi is really not a convenient option. Side-stream treatment where pumping is already in place is an ideal way to introduce the use of venturis to create small bubbles. In particular, consider using the type of venturis used with hot tubs and spa baths. These units are usually very economical as well.

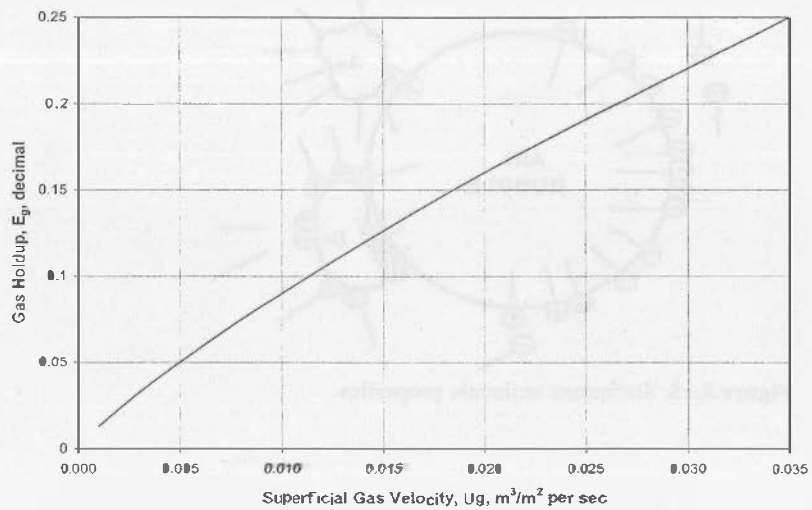


Figure 5.17 Gas holdup, E_g, versus superficial gas velocity, U_g.

SIZING FOAM FRACTIONATORS

While it is not really possible to exactly predict foam fractionator performance, the findings of Weeks et al. (1992) show that the removal rates of protein were independent of water flow but were directly proportional to airflow. In other words, it is the quantity of air bubbles that was the important parameter. So, using some gross approximations, the volatile solids (VS, an approximate measure of organic matter) removal rate can be made proportional to the air flow rate used in the fractionator column (units do not balance):

$$R_{vs} = 0.40 \cdot Q_{air,Lpm} \quad (5.13)$$

Equation 5.13 is an approximation to the Weeks et al. (1992) model and assumes that the foam fractionator is well designed and follows the general guidelines previously given for maximizing performance of a unit. It was developed from data for waste water that approximately had 300 ppm VS, 10 ppm TSS and 0.8 ppm TKN in the fish culture tank. Correlations and enrichment factors for volatile solids (VS), TSS and Total-Kjeldahl-Nitrogen (TKN) (E_{VS}, E_{TSS}, and E_{TKN}) were also developed for the foam condensate obtained from these waters using an airlift type fractionator:

$$TSS = 0.27\ VS \quad (5.14)$$

$$TKN = 0.038\ VS \quad (5.15)$$

$$E_{VS} = 2.7 \text{ (307 increased to 816 } ppm) \quad (5.16)$$

$$E_{TSS} = 25 \text{ (10 increased to 251 } ppm) \quad (5.17)$$

$$E_{TKN} = 44 \text{ (0.8 increased to 34.6 } ppm) \quad (5.18)$$

EXAMPLE PROBLEM

How much airflow is required for foam fractionation for 10,000 fish @ 1 kg fed 1% of their body weight per day?

<u>Solution</u>.

TSS generated (Rule of thumb)

$$= 0.25 \frac{g_{TSS}}{g_{Feed}} \cdot \frac{0.01 g_{feed}}{g_{fish} \cdot day} \cdot \frac{1{,}000 g_{fish}}{fish} \cdot 10{,}000 fish$$

$$= 25{,}000 \frac{g_{TSS}}{day}$$

Assume 3% of the TSS is generated as fine solids (<30 μm) that are to be removed by the fractionators. Rearranging Eq. 5.13 to solve for the required airflow and assuming that all the TSS are VS:

$$Q_{air,Lpm} = \frac{R_{VS}}{0.4} = 0.03 \cdot \frac{\left(\frac{25{,}000 g}{day}\right)}{0.4} = 1{,}875 Lpm\ air\ flow$$

From Fig. 5.17, the superficial gas flow, U_g, for a fractionator column expanded 25% (for maximum effectiveness), shows a required U_g of 0.035 m^3/m^2 per sec or converting to Lpm per cm^2

$$U_g = 0.035 \cdot \frac{m^3/m^2}{s} \cdot 6 \frac{\frac{Lpm}{cm^2}\ air\ flow}{\frac{m^3/m^2}{s}} = 0.21 \frac{Lpm}{cm^2}\ air\ flow$$

Required cross sectional area of the foam fractionator units

$$A = \frac{1{,}875\ Lpm\ air\ flow}{0.21 \frac{Lpm}{cm^2}} = 8{,}930\ cm^2$$

These are the numbers for 100 kg of feed being fed, so we can express these requirements per kg of feed fed per day:

$$\frac{Q_{air,Lpm}}{\frac{kg\ feed}{day}} = \frac{19\ Lpm\ air\ flow}{\frac{kg\ feed}{day}} \cong 20\ Lpm\ air\ flow\ (roundup)$$

$$\frac{A}{\frac{kg\ feed}{day}} = 89\ cm^2 \cong 90\ cm^2\ (roundup)$$

And this gives us another rule of thumb (rounding the above numbers) for foam fractionators:

> **"Rule of Thumb"**
> **<u>Foam Fractionators</u>**
> Per kg of feed fed per day (assumes 3% of TSS is quantity of fine solids to be removed by fractionation)
> - 20 Lpm of air flow
> - 90 cm^2 of column cross sectional area

If airlift type fractionators are used, these calculations would determine that a 15 cm diameter fractionator is required for each 2 kg of feed fed per day. This is consistent with the author's observations on an intensive trout operation that was discharging only 3% of system volume per day; the water was dark but very clear.

Finally, return to our earlier example of the amount of protein being generated from dead fish in a tank (350 g protein per day). Chen et al. (1994b) determined that only 1 mg of protein existed in 331 mg of VS collected as foam condensate. Doing the reverse math, the 350 g of protein (which effectively could become all surfactant material) would provide foaming potential for the following quantity of TSS or VS:

$$\theta_{TSS} = (350\ g_{protein}) \cdot 331 \frac{g_{VS}}{g_{protein}} = 115{,}850\ g_{VS}$$

Assuming that 100% of the VS is TSS type organic material, then this quantity of TSS material would require about the following number of days for a foam fractionator process to remove these TSS (based upon the previous calculation):

$$\frac{115{,}850\ g_{TSS}}{25{,}000 \frac{g_{TSS}}{day}} = 4.6\ days$$

Thus, you might expect to see this tank under these conditions foam excessively for 2 to 3 days and it would be 4 or 5 days before the tank returned to normal foam levels.

5.6 Design Example – Solids Capture

As previously described, Omega Industries (OI) requires an engineering plan for the construction and operation of an Omega Fish Aquaculture Facility (OFAF). The production strategy is a three stage: juvenile, fingerling and growout stages. Table 5.7 summarizes the design so far, consisting of the tank biomass, feed rates, and tank volume. Flow rates were estimated at this point based on either required tank exchanges per hour or that which is needed for either dissolved oxygen or ammonia-nitrogen removal.

As a rule of thumb, 25% to 35% of the feed fed to the fish will be excreted as suspended solids (or total suspended solids, TSS) on a dry matter basis. As previously discussed in Chapter 4, the use of the Cornell dual-drain system will result in a 10-fold higher concentration of TSS from the center drain to the influent water to the solids capture device. In this design example, it will be assumed that 20 to 25% of the flow will be from the center drain and will contain the majority of the suspended solids.

Table 5.7 Final Biomass, Feed Rates, Tank Volume and Flow Rates of the Three Stage Omega Fish Production Strategy

	Tank Biomass kg (lbs)	Feed Rate per day	Volume m^3 (gal)	Flow Rate Lpm(gpm)
Juvenile:	193 kg (425 lbs)	3.0 kg (6.6 lbs)	6.4 m^3 (1694 gal)	321 Lpm (85 gpm)
Fingerling:	450kg (990 lbs)	5.7 kg (12.6 lbs)	11.3 m^3 (2978 gal)	379 Lpm (100 gpm)
Growout:	875 kg (1925 lbs)	9.6kg (21.1 lbs)	17.5 m^3 (4621 gal)	379 Lpm (100 gpm)

Since it is somewhat difficult to follow the biomass loading and feeding rates, a simple spreadsheet was used to calculate the biomass and feed rate for each of the three growout stages and then combine the appropriate cells to yield final biomass and feed rate per day values that would be associated with a pod and life support system (LSS). These are summarized in Table 5.8, 5.9, and 5.10 for the juvenile, fingerling and growout Stages.

The authors often disagree on details of design and this is one of those cases. One author would utilize two separate LSS's for the juvenile production stage (five tanks per LSS) and five separate Life Support System (LSS) for the fingerling/growout stages (two fingerling and two growout tanks per LSS). Each of the tanks in the two juvenile production pods would be stocked out at a two week interval. The logic to this approach is that if there were to be a total failure of one juvenile pod, production would be reduced by half, but still fish would be available every two weeks, instead of every week. Five LSS's would be used in the fingerling/growout stages by combining two fingerling and two growout systems into a pod and supported by its own LSS. Both the fingerling and the growout tanks in each pod would be stocked out at a five week interval (½ cycle stocking), thus reducing the total biomass, daily feeding rate, and thus the size of the required LSS. This would also significantly reduce losses due to failure of one pod's LSS or other catastrophic failures.

The other author believes that the fewest LSS's possible should be used and in this case would have a single LSS for the juvenile production stage and two LSS's each supporting a pod with a combination of five fingerling and five growout tanks. To help prevent catastrophic loss of the juvenile fish, the stocking density in the juvenile system would be reduced to 20 kg/m^3, allowing the fish to survive loss of the pod's LSS with only in tank aeration. With two LSS's, only half of the production would be lost if there was a catastrophic failure of the LSS and with an alternating stocking sequence, the harvest would be reduced to every two weeks, instead of every week, thus maintaining at least some consistency (~50% of intended) in production over the year.

Table 5.8 Juvenile Stage, Biomass (kg) and Daily Feed Rate (kg/day).

Week	Length (inches)	Weight (gms)	Biomass (kg)	% bw/day	Feed Rate (kg/day)
0	5.25	50.0	58.3	1.91%	
1	5.51	57.7	67.2	1.88%	1.26
2	5.77	66.1	77.1	1.85%	1.42
3	6.02	75.4	87.8	1.81%	1.59
4	6.28	85.4	99.6	1.78%	1.77
5	6.54	96.3	112.3	1.74%	1.96
6	6.79	108	126.0	1.71%	2.15
7	7.05	121	140.9	1.67%	2.36
8	7.31	135	156.8	1.64%	2.57
9	7.56	149	174.0	1.60%	2.79
10	7.82	165	192.3	1.56%	3.01
				Total Feed:	20.9

Table 5.9 Fingerling Stage, Biomass (kg) and Daily Feed Rate (kg/day).

Week	Length (inches)	Weight (gms)	Biomass (kg)	Feed Rate % bw/day	Feed (kg/day)
11	8.08	182	212	1.54%	3.25
12	8.33	200	233	1.51%	3.51
13	8.59	219	255	1.48%	3.77
14	8.85	239	278	1.45%	4.04
15	9.10	260	303	1.42%	4.32
16	9.36	283	330	1.39%	4.60
17	9.62	307	358	1.37%	4.89
18	9.87	332	387	1.34%	5.18
19	10.13	359	418	1.31%	5.48
20	10.39	387	451	1.28%	5.78

Table 5.10 Growout Stage, Biomass (kg) and Daily Feed Rate (kg/day).

Week	Length (inches)	Weight (gms)	Biomass (kg)	Feed Rate % bw/day	Feed (kg/day)
21	10.64	416	485	1.26%	6.13
22	10.90	447	521	1.25%	6.50
23	11.16	479	558	1.23%	6.87
24	11.41	513	598	1.21%	7.26
25	11.67	548	639	1.20%	7.65
26	11.93	585	682	1.18%	8.05
27	12.18	624	727	1.16%	8.46
28	12.44	664	774	1.15%	8.87
29	12.70	706	823	1.13%	9.29
30	12.95	750	874	1.11%	9.72

For the first design scenario with two LSS's for the juvenile stage, the total feed rate per day with alternate weekly stocking in five production tanks per juvenile pod is a maximum of 10.9 kg (24 lbs) with a maximum biomass of 652 kg (1434 lbs) per system as shown in Table 5.11. In this scenario, the juvenile system is stocked at a density 30 kg/m^3 (0.25 lbs/gal). The corresponding design tank size is 3.28 m (10 ft) in diameter with a volume of 6.4 m^3 (1693 gallons). Since there are 5 tanks per juvenile pod, the total pod volume would be approximately 32.0 m^3 (8,465 gallons). Assuming a tank exchange rate of 20 minutes (to maximize system water quality) would mean a flow rate through the

system of about 90.8 m^3/hr (400 gpm). This flow would be partitioned between a center drain discharge (25% of the flow) of 22.7 m^3/hr (100 gpm) and a sidewall discharge (75% of the flow) of 68.1 m^3/hr (300 gpm).

Table 5.11. Daily Feed Rate (kg/day) and Total Feed Rate (kg) for Two Juvenile Pods (supported by LSS's), Each Stocked at Two Week Intervals (each juvenile pod system consists of 5 tanks)

Week	Feed (kg)	Feed (kg) LSS #1	LSS#2
1	1.26	1.26	
2	1.42		1.42
3	1.59	1.59	
4	1.77		1.77
5	1.96	1.96	
6	2.15		2.15
7	2.36	2.36	
8	2.57		2.57
9	2.79	2.79	
10	3.01		3.01
	Total Feed:	**10.0**	**10.9**

In the first design scenario, five pods and associated LSS's are used for the fingerling/growout stages consisting of two fingerling tanks and two growout tanks per pod and the corresponding daily feed rate and total feed rates for each system is summarized in Table 5.12.

Table 5.12 Daily Feed Rate (kg/day) and Total Feed Rate (kg) for Each of Five Pods (supported by LSS's), Stocked at Five Week Intervals (each pod consists of two fingerling and two growout tanks).

Week	Fingerling Feed/day (1/2 Cycle) (kg)		Week	Growout Feed/day (1/2 Cycle) (kg)		POD Total (kg)
11 & 16	3.25	4.60	21 & 26	6.13	8.05	22.0
12 & 17	3.51	4.89	22 & 27	6.50	8.46	23.3
13 & 18	3.77	5.18	23 & 28	6.87	8.87	24.7
14 & 19	4.04	5.48	24 & 29	7.26	9.29	26.1
15 & 20	4.32	5.78	25 & 30	7.65	9.72	**27.5**

Thus the maximum daily feed rate for each of the five LSS's for the fingerling/growout stages is 27.5 kg per day (60.5 lbs/day) and the maximum biomass in the each system is 2267 kg (5,000 lbs).

The fingerling tanks are 3.65 m (12 ft) in diameter with a volume of 11.3 m^3 (2978 gallons). Based on the required flow rates for oxygen, TAN, carbon dioxide, TSS or Tank Exchange, Table 3.5, the flow rate for one fingerling tank is 22.7 m^3/hr (100 gpm). In this case, the defining criteria for flow rate was based on a tank hydraulic retention time (HRT) or the time to exchange on tank volume of 30 minutes. The growout tanks are 4.57 m (15 ft) in diameter with a volume of 17.5 m^3 (4620 gallons). Again based on the required flow rates for oxygen, TAN, carbon dioxide, TSS or Tank Exchange, Table 3.5, the flow rate for one growout tank was determined to be 22.7 m^3/hr (100 gpm). This value was also defined by the HRT criteria of 45 minutes chosen for growout. Thus the total exchange rate for a POD of two fingerling and two growout tanks accumulates to 90.8 m^3/hr (400 gpm). Using the Cornell dual drain approach, this flow would be partitioned between a center drain discharge (25% of the flow) of 22.7 m^3/hr (100 gpm) and a sidewall discharge (75% of the flow) of 68.1 m^3/hr (300 gpm).

For the second design scenario with a single LSS for the juvenile stage, the total feed rate per day is 20.9 kg (46 lbs) with a biomass of 1234 kg (2715 lbs) as shown in Table 5.8 or add the two feeding loads shown in Table 5.11, since there is only one LSS and one Pod. (Note: in Table 5.8, there are 10 cohorts or ten weeks of cohort placements all being supported by the same LSS, and this is how we arrive at the 20.9 kg/day of feeding). In order to minimize impact of catastrophic system failure, the tank stocking density in this scenario is reduced to 20 kg/m^3 (0.166 lbs/gal). This would result in an increase in tank size to 3.66 m (12 ft) in diameter with a volume of 9.61 m^3 (2540 gallons). Since there are 10 tanks, the total system volume would be approximately 96.1 m^3 (25,400 gallons). Assuming a tank exchange rate of 20 minutes (to maximize system water quality) would mean a flow rate through the system of about 273 m^3/hr (1200 gpm). This flow would be partitioned between a center drain discharge (25% of the flow) of 68.3 m^3/hr (300 gpm) and a sidewall discharge (75% of the flow) of 205 m^3/hr (900 gpm).

In the second design scenario, two LSS are used for the fingerling/growout stages and the corresponding daily feed rate and total feed rates for each system is summarized in Table 5.13. Based on this table, the maximum feed rate for the two LSS for the fingerling/growout stages is 63.5 kg per day (140 lbs/day) and the maximum biomass in the each system is 5128 kg (11,280 lbs). The fingerling tanks are 3.65 m (12 ft) in diameter with a volume of 11.3 m^3 (2978 gallons). Based on the

required flow rates for oxygen, TAN, carbon dioxide, TSS or Tank Exchange, Table 3.5, the flow rate for one fingerling tank is 22.7 m^3/hr (100 gpm). In this case, this was based on a tank hydraulic retention time (HRT) or the time to exchange on tank volume of 30 minutes. The growout tanks are 4.57 m (15 ft) in diameter with a volume of 17.5 m^3 (4620 gallons). Again based on the required flow rates for oxygen, TAN, carbon dioxide, TSS or Tank Exchange, Table 3.5, the flow rate for one growout tank was determined to be 22.7 m^3/hr(100 gpm). This value was also based on an HRT of 45 minutes for growout. Thus the total exchange rate for a POD of five fingerling and five growout tanks is 227 m^3/hr (1000 gpm). This flow would be partitioned between a center drain discharge (20% of the flow) of 45.4 m^3/hr(200 gpm) and a sidewall discharge (80% of the flow) of 181.7 m^3/hr(800 gpm).

Table 5.13. Daily Feed Rate (kg/day) and Totals (kg) for Two LSS, Fingerling & Growout, Stocked at Two Week Intervals.

Week	Fingerling LSS #1	Fingerling LSS #2 (kg)	Growout LSS #1	Growout LSS #2 (kg)	Single Pod LSS #1	Single Pod LSS#2 (kg)
11&21	3.25	---	6.13	---	9.39	
12&22	---	3.51	---	6.50	---	10.0
13&23	3.77	---	6.87	---	10.6	
14&24	---	4.04	---	7.26	---	11.3
15&25	4.32	---	7.65	---	12.0	
16&26	---	4.60	---	8.05	---	12.6
17&27	4.89	---	8.46	---	13.3	
18&28	---	5.18	---	8.87	---	14.5
19&29	5.48	---	9.29	---	14.8	
20&30	---	5.78	---	9.72	---	15.49
Total:	21.7	23.1	38.4	40.4	60.1	63.5

Table 5.14. Summary of Design Total Volume and Design Flows for the Two Design Scenarios

	Design Scenario One: Two Juvenile/Fry Pods	Design Scenario One: Five Fingerling/ Growout Pods	Design Scenario Two: Single Juvenile Pod	Design Scenario Two: Two Fingerling/ Growout Pods
Pod Total Volume:	32.0 m^3 (8,465 gal)	57.2 m^3 (15,110 gal)	96.1 m^3 (25,400 gal)	143.8 m^3 (38,000 gal)
Total Flow:	90.8 m^3/hr (400 gpm)	90.8 m^3/hr (400 gpm)	273 m^3/hr (1200 gpm)	227 m^3/hr (1000 gpm)
Center Discharge:	22.7 m^3/hr (100 gpm)	22.7 m^3/hr (100 gpm)	68.3 m^3/hr (300 gpm)	45.4 m^3/hr (200 gpm)
Side-wall Discharge:	68.1 m^3/hr (300 gpm)	68.1 m^3/hr (300 gpm).	205 m^3/hr (900 gpm)	181.7 m^3/hr (800 gpm)
Feed Rate Per day:	10.9 kg (24 lbs)	27.5 kg (60.5 lbs)	20.9 kg (46 lbs)	63.5 kg (140 lbs)

Swirl Separator/ Radial-flow Separator

Since the fecal matter from the Omega Fish are relatively large and intact (personal observation), a swirl separator or radial-flow settler before the microscreen filter could significantly reduce the total loading on the solids capture system. Davidson and Summerfelt (2004) reported solids removal efficiencies for the discharge from the center drain to a radial-flow settler at low and high feed loading rates of 72.1 and 79.5%, respectively. By pretreating the center drain discharge with a radial-flow settler, a significant reduction in the loading on the downstream treatment system would be seen. Designs often used in the salmonid industry combine the treated discharge flow (overflow from the radial flow separator with the lower solids concentration) with the side-wall discharge from the production tank and then the combined flow is treated with a single microscreen filter for optimal solids capture (to ensure any solids that are escaping from the radial flow separator are captured). The surface-loading rate applied to the radial-flow settler used by Davidson and Summerfelt (2004) was 0.0031 m^3/s per square meter (4.6 gpm/ft^2) of settling area.

As an example, for the first design scenario of five juvenile tanks on a single LSS, a 22.7 m^3/hr (100 gpm) center drain discharge would require 2.0 m^2 (21.7 ft^2) of swirl/radial flow separator cross sectional area, which could be provided using a ~1.6 m (5.2 ft) diameter radial-flow separator. For the second design scenario of a single LSS for ten juvenile tanks, a flow of 68.3 m^3/hr (300 gpm) from the center drains, would require 6.1 m^2 (65.2 ft^2) settling area, which can be provided using a 2.8 m (9.1 ft) diameter radial-flow separator. Radial-flow separators are commercially available from several fiberglass tank manufacturers. For example, Marine Biotech (division of Aquatic Habitats, Apopka, FL, see www.aquatichabitats.com) manufactures several models of radial flow separators with diameters from 12 inches to 132 inches. Based on their technical data specifications, a 60 inch radial flow separator will treat a design flow of 28.4 m^3/hr (125 gpm) and a 120 inch diameter a flow rate of 112.5 m^3/hr (495 gpm).

Microscreen Filters

Microscreen filters for filtration are popular because they require minimal labor and floor space in comparison to a settling basin. Screen filters remove solids by virtue of physical restrictions (or straining) on a media when the mesh size of the screen is smaller than the particles in the wastewater. Microscreen filters are designed to treat large flow rates and their capital costs puts them at a disadvantage at lower flow rates compared to some other options. Thus the microscreen filter would be appropriate for the higher flow rates, but other solids capture devices such as a propeller-washed bead filters (Figure 5.14) might be more appropriate for the lower discharge rates.

The design process is very straight forward, just consult the manufacturers spec sheets, and find the correct filter that corresponds to the system parameters in your application. Specifications are often divided into cold water and warm water systems and expected intake TSS concentration. For example, for the first design scenario of two fingerling/growout tanks on a single LSS with a 22.7 m^3/hr (100 gpm) center drain discharge and warm water conditions, a Hydrotech (see Table 5.5, www.hydrotech.se) Model 501 would be required, with a design flow of 21.6 m^3/hr (95 gpm; Table 5.5 shows capacity at 6 L/s or 95 gpm maximum, so at the unit's maximum capacity) with a 60 micron screen filter. This would suggest one of the smallest microscreens manufactured with a corresponding high capital cost compared to the higher flow rate for the single LSS design. For the second design scenario of five fingerling/growout tanks on a single LSS, a flow of 45.4 m^3/hr (12.6 L/s, or 200 gpm) from the center drains would require a Model 801 would be suggested with a design flow rate of 54 m^3/hr (15 L/s, or 238 gpm) with a 60 micron screen filter (the Hydrotech Model 802 would handle up to 158 m^3/hr or 697 gpm).

If a radial-flow separator is used to pre-treat the discharge water then often both the center drain and the side-wall discharges are combined and treated via a microscreen filter. In the case of a Hydrotech microscreen filter, the filter flow through rate would then be adjusted to correspond to a smaller solids content. Thus for example for the first design scenario a total flow of 90.8 m^3/hr (25.2 L/s, 400 gpm) would require treatment and a Model 802 could be used with a design flow rate of 112 m^3/hr (31 L/s, or 490 gpm) with a 60 um screen filter. In the second design scenario, the total flow for treatment would be 273 m^3/hr (75.8 L/s or 1200 gpm) and a Model 1603 with a design flow rate of 331 m^3/hr (92 L/s or 1460 gpm) with a 60 micron screen filter.

Granular Media (GM) Filters

Granular media filtration involves passage of water through a bed of granular material (media) resulting in the deposition of solids onto the media. One of the more popular types is the Propeller-washed bead filter manufactured by Aquaculture Systems Technologies, LLC (www.beadfilters.com), which is available in several different sizes based on media volume (Table 5.15). These are sized either as bioclarifiers (solids removal and biofiltration) or simple as solids capture devices, which will accept high loading rates for the same sized unit. As a solids capture device or clarifier, the PBF is sized according to either the maximum daily feed input or one that is compatible with the biological filters design flow rate. For solids capture only, the design value is the device will collect solids being generated from a feed loading of 96 kg feed/m^3 of media (6 lbs/ft^3) (note there are no time units here, but it is however long it takes to account for this quantity of feed being fed to the system).

Thus for the first design scenario, the maximum feed rate for one of the juvenile production pods is 10.9 kg (24 lbs) of feed per day, thus approximately 0.11 m^3 (4 ft^3) of media would be required for solids capture only. This could be accomplished with PBF-5S, with a design feed rate of approximately 13.2 kg (30 lb) of feed per day and a maximum flow rate of 22.7 m^3/hr (100 gpm). For the two fingerling/growout tanks on a single LSS with a 27.5 kg (60.5 lbs) daily feed rate a PBF-10 would handle the solids loading at a maximum flow rate of 22.7 m^3/hr (100 gpm).

Finally for the second design scenario, the maximum feed rate for the juvenile production pods is 20.9 kg (46 lbs) of feed per day, thus

approximately 0.22 m^3 (8 ft^3) of media would be required for solids capture only. This could be accomplished with PBF-25S, with a design feed rate of approximately 56.8 kg (125 lb) of feed per day and a maximum flow rate of 68.1 m^3/hr (300 gpm). For one of the two fingerling/growout pods with a 63.5 kg (140 lbs) daily feed rate a PBF-25 would handle the solids loading at a maximum flow rate of 45.4 m^3/hr (200 gpm).

Table 5.15. Summary of Propeller-Washed Bead Filter Design Recommendations

PBF Model	Media Volume ft^3 (m^3)	Max. Flow Rate, gpm (Lpm)	Solids Only, lbs feed/day (kg/day)
PBF-3	3(0.08)	30(110)	15(6.81)
PBF-5	5(0.14)	50(190)	25(11.3)
PBF-5S		100(380)	
PBF-10	10(0.28)	100(380)	50(22.7)
PBF-10S		200(760)	
PBF-25	25(0.70)	200(760)	125(60.7)
PBF-25S		400(1500)	

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LIST OF SYMBOLS

a_{TSS}	TSS produced as a proportion of feed fed (kg TSS per kg feed)
A	Cross sectional area of the column, m^2
A_{sz}	Settling basin floor area (settling zone only), m^2
BOD_5	Biochemical Oxygen Demand 5 day, mg/L
C_D	Drag coefficient, dimensionless
D	Depth of sedimentation basin, m
D_p	Diameter of particles, m
Eg	Gas holdup (fraction)
E_{VS}	Enrichment factors for volatile solids
E_{TSS}	Enrichment factors for TSS
E_{TKN}	Enrichment factors for TKN
f_{rem}	fraction of solids removed
F	Settling zone safety factor
g	Gravitational acceleration, m/s^2
o.d.	Outside diameter of a particle or other object, length unit
P_{TSS}	TSS production rate (kg TSS produced per day)
Q	Water flow, m^3/time
$Q_{air,Lpm}$	Air flowrate, Lpm
Q_{air}	Air flow rate through column, m^3/s
Q_{out1}	Flow rate leaving the sidewall drain (m^3 per day)
Q_{out2}	Flow rate leaving the bottom center drain (m^3 per day)
r_{feed}	Feeding rate (kg feed per kg fish per day)
R_{vs}	Removal rate of volatile solids, g/day
SS	Suspended solids, mg/L
TKN	Total-Kjeldahl-Nitrogen, mass
TP	Total phosphorus, mg/L
TSS	Total suspended solids, mg/L
$TSS_{capture\,\%}$	Percent removed of incoming TSS, %
TSS_{in}	TSS concentration entering unit (kg/m^3 or ppm)
TSS_{out1}	TSS concentration leaving the sidewall drain (kg/m^3)
TSS_{out2}	TSS concentration leaving the bottom center drain (kg/m^3)
VS	Volatile solids, mg/L
V_{basin}	Basin volume, m^3
V_o	Volumetric flow of water per unit surface area, m^3/m^2 time
V_{tank}	Volume of water contained within culture unit (m^3 culture volume)
V_s	Velocity of settling particles, m/s
U_b	Bubble rising velocity, m/s

U_g	Superficial gas velocity, m/s
ρ_{fish}	Density of fish in the culture tank (kg fish per m^3 culture volume)
ρ_p	Density of particles, kg/m^3
ρ	Density of water, kg/m^3
τ	Hydraulic retention time
μ	Dynamic viscosity, Pa·sec.
Θ_{TSS}	Mass of protein generated for foaming, g

CHAPTER 6

WASTE MANAGEMENT & DISPOSAL

6.0 INTRODUCTION

As environmental regulations become more stringent environmentally sound waste management and disposal will increasingly be more important in all agriculture/aquaculture operations, particularly marine aquaculture. Because of their high moisture content, the management of solid wastes generated by aquaculture systems presents unique storage and disposal problems. Two of the primary concerns are the suspended solids and phosphorous in the discharged effluent. Mechanical filters such as microscreens, sand filters, and floating bead filters generate a separate solids waste effluent stream discharge that benefits from further concentration to reduce the quantity and improve the quality of discharge. These sludge processing overflows are often combined with waters spilled or flushed from the RAS to form the discharge (effluent) from the facility. For marine RAS it is critical that a significant percentage of the waste effluent stream be recycled back into the production system to conserve salts and reduce and/or eliminate the environmental impact of salt discharges. In addition, thickening wastes through dewatering increases the options available for final disposal and reduces the volume and costs of storage and transportation. Development of improved systems for the disposal and utilization of aquaculture wastes are critical priorities, as enhanced waste management systems will improve the economic viability and sustainability of aquaculture systems. Optimally, aquaculture wastes should be utilized as an environmentally beneficial product.

A proper waste management strategy is now considered critical for maintaining the legality, profitability, and sustainability of any aquaculture facility. The U.S. Environmental Protection Agency (US EPA) completed a major study to determine if and how aquacultural waste discharges should be regulated (see full text of effluent guidelines at end of chapter). This is an ongoing assessment, which could have a major impact on the industry. One of the problems that EPA faced was the wide range of species, technologies, water sources, etc. that are encountered in the aquaculture industry. Recirculating systems can play a leading role in the future of intensive aquaculture systems, due to their

limited water usage and an easily treated low volume, high concentrated waste stream.

Congress passed the Federal Water Pollution Control Act (1972), also known as the Clean Water Act (CWA), to "restore and maintain the chemical, physical, and biological integrity of the Nation's waters." The CWA establishes a comprehensive program for protecting our nation's waters. Among its core provisions, the CWA prohibits the discharge of pollutants from a point source to waters of the U.S. except as authorized by a National Pollutant Discharge Elimination System (NPDES) permit. The CWA also requires EPA to establish national technology-based effluent limitations guidelines and standards (effluent guidelines or ELG) for different categories of sources, such as industrial, commercial, and public sources of waters. Effluent guidelines are implemented when incorporated into an NPDES permit. Effluent guidelines can include numeric and narrative limitations, including Best Management Practices, to control the discharge of pollutants from categories of point sources.

The EPA issues the National Pollution Discharge Elimination System (NPDES) permit to regulate various pollutants from point sources, including aquaculture. NPDES permits are required for fish hatcheries, fish farms, or any other facilities that raise aquatic animals under the following conditions:

1. Coldwater fish species or other coldwater aquatic animals in ponds, raceways, or similar structures that discharge at least 30 days per year, produce more than 9,090 kg (20,000 lbs) of aquatic animals per year, or receive more than 2,273 kg (5,000 lbs) of food during the month of maximum feeding.

2. Warmwater fish species or other warmwater aquatic animals in ponds, raceways, or similar structures that discharge at least 30 days per year. This does not include closed ponds, which discharge only during periods of excess run-off, or warmwater facilities, which produce less than 45,454 kg (100,000 lb) of aquatic animals per year.

3. Facilities determined on a case-by-case basis by the permitting authority to be significant contributors of pollution to waters of the United States. Discharge of pollutants to receiving waters from aquaculture production facilities, except as provided in the permit, is a violation of the Clean Water Act and may be subject to enforcement by EPA.

NPDES permits for aquaculture operations can set discharge limits on solids, nutrients, and chemical compounds used for water treatments. A provision under the CWA allows the EPA to transfer or "delegate" its NPDES permit authority to individual states to regulate point-source discharges into waters located within their borders, commonly referred to as "waters of the state." To become a delegated state, resource agencies must submit a regulatory plan to the EPA for approval and demonstrate that state laws provide adequate legal authority to carry out the program described. State programs must be equivalent to the EPA's and may impose requirements that are more stringent. Of those states receiving delegated status, most choose to incorporate the NPDES permit into their own regulatory program by issuing a joint state/federal permit.

6.1 EPA Effluent Limitation Guidelines - 2004

Final Effluent Limitation Guidelines - 2004

On September 22, 2004, the EPA released its Effluent Limitations Guidelines and New Source Performance Standards for the Concentrated Aquatic Animal Production Point Source Category, 40 CFR Part 451. This was after two years of continued review, site visits, and representative water quality testing of several aquaculture facilities, and additional public, NGO and state and federal agency comments. Several sections of the introductory text are included here to show the background of the final guidelines.

40 CFR Part 451

Effluent Limitations Guidelines and New Source Performance Standards for the Concentrated Aquatic Animal Production Point Source Category

> **SUMMARY:** Today's final rule establishes Clean Water Act effluent limitations guidelines and new source performance standards for concentrated aquatic animal production facilities. The animals produced range from species produced for human consumption as food to species raised to stock streams for fishing. The animals are raised in a variety of production systems. The production of aquatic animals contributes pollutants such as suspended solids, biochemical oxygen demand, and nutrients to the aquatic environment. The regulation establishes technology-based narrative limitations and standards for wastewater discharges from new and existing concentrated aquatic animal production facilities that discharge directly to U.S. waters. EPA estimates that compliance with this regulation will affect 242 facilities. The rule is projected to reduce the discharge of total suspended solids by about 0.5 million pounds per year and reduce the discharge of biochemical oxygen demand

(BOD) and nutrients by about 0.3 million pounds per year. The estimated annual cost for commercial facilities is $0.3 million. The estimated annual cost to Federal and State hatcheries is $1.1 million. EPA estimates that the annual monetized environmental benefits of the rule will be in the range of $66,000 to $99,000.

Subpart B—Recirculating Systems

§ 451.20 Applicability

This subpart applies to the discharge of pollutants from a concentrated aquatic animal production facility that produces at least 100,000 pounds a year in flow-through and recirculating systems that discharge wastewater at least 30 days a year (used primarily to raise trout, salmon, hybrid striped bass and tilapia).

TSS LIMITATIONS:

EPA's decision to not establish national numeric limits for TSS will not restrict a permit writer's authority to impose site-specific permit numeric effluent limits on the discharge of TSS or other pollutants in appropriate circumstances. For example, a permit writer may establish water quality-based effluent limits for TSS (see 40 CFR 122.44(d)) or regulate TSS (by establishing numeric limits) as a surrogate for the control of toxic pollutants (see 40 CFR 122.44(e)(2)(ii)) where site-specific circumstances warrant. The permit writer may also issue numeric limits in general permits applicable to classes of facilities. In fact, one of the bases for EPA's decision not to establish uniform national TSS limits is the recognition that a number of states, particularly those with significant numbers of CAAP facilities, already have general permits with numeric limits tailored to the specific production systems, species raised, and environmental conditions in the state, and these permits seem to be working well to minimize discharges of suspended solids (see DCN 63056). EPA believes there would be minimal environmental gain from requiring these states to redo their General Permits to conform to a set of uniform national concentration-based limits that in most cases would not produce significant changes in control technologies and practices at CAAP facilities.

EPA's existing National Pollutant Discharge Elimination System (NPDES) regulations define when a hatchery, fish farm, or other facility is a concentrated aquatic animal production facility and, therefore, a point source subject to the NPDES permit program. See

40 CFR 122.24. In defining "concentrated aquatic animal production (CAAP) facility," the NPDES regulations distinguish between warmwater and coldwater species of fish and define a CAAP facility by, among other things, the size of the operation and frequency of discharge. A facility is a CAAP facility if it meets the criteria in 40 CFR 122 appendix C or if it is designated as a CAAP facility by the NPDES program director on a case-by-case basis. Today's action does not revise the NPDES regulation that defines CAAP facilities.

Although the new national effluent regulations do not contain a numerical limit for TSS and other water quality parameters, it is important to note that many states aquaculture waste disposal policies are set by the individual states through NPDES permits. The level of development of state policy largely is a function of the amount and scale of industry activity and the experience of regulatory agency staff. States employ a variety of classification schemes but generally include categories equivalent to protection of public water supply; fish, wildlife and aquatic life; primary contact recreation (swimming); secondary contact recreation (boating); agricultural water supply; and industrial water supply. Most states designate water bodies for multiple uses. Water quality criteria are expressed in narrative form and/or as a list of water quality parameters and aesthetic values such as temperature, pH, dissolved oxygen, nitrogen, phosphorous, sedimentation, coliform bacteria, oil and grease, color, turbidity, slicks, odors, surface floating solids, and radioactive and toxic substances (Ewart et al., 1995).

State water quality standards also contain an "antidegradation" statement designed to maintain and protect existing uses. Special provisions, however, may be included for protection of waters of exceptionally high quality or those considered high priority waters based upon aesthetic, ecological, recreational, or other factors. This generally means that any approved discharges must be of equal or better quality than the receiving waters or that all new discharges are prohibited (Ewart et al., 1995).

In addition the new rule requires that all applicable facilities:

- Prevent discharge of drugs and pesticides that have been spilled and minimize discharges of excess feed.
- Regularly maintain production and wastewater treatment systems.
- Keep records on numbers and weights of animals, amounts of feed, and frequency of cleaning, inspections, maintenance, and repairs.
- Train staff to prevent and respond to spills and to properly operate and maintain production and wastewater treatment systems.

- Report the use of experimental animal drugs or drugs that are not used in accordance with label requirements.
- Report failure of or damage to a containment system.
- Develop, maintain, and certify a Best Management Practice plan that describes how the facility will meet the requirements.

6.2 WASTE MANAGEMENT

Handling wastewater is a major problem in all animal agriculture systems, but there are substantial differences between aquaculture wastewater and manure from hog or dairy systems. The latter are typically in the range of 5 to 15% suspended solids, while fish wastewater can be anywhere from 0.2 to 4.0% suspended solids (Twarowska et al. 1997; Bergheim et al. 1993; Cripps & Bergheim, 2000). Typical suspended solids concentrations from drum filters used in intensive aquaculture operations are around 0.5% (Ebeling and Summerfelt, 2002). Suspended solids are captured in a variety of ways, but capture techniques primarily rely on straining, settling, or a combination of both. These systems are reasonably effective for removing suspended solids from the culture water, but require periodic flushing of the device to maintain removal efficiency between flushings. Water lost in the flushing process must of course be replaced with clean or new makeup water.

The large flow rates involved in maintaining water quality and removing suspended solids can result in a significant cumulative waste load discharge from fish farms (Braaten, 1991; IDEQ, 1998; Summerfelt, 1999). Consistently meeting strict discharge standards can also be difficult because pipe, channel, and tank cleaning routines can produce fluctuations in discharge flowrates and in the consistencies and concentrations of waste material. The distribution of the nutrients and organic matter between the dissolved, suspended, and settleable fractions affects the choice of method used and the difficulty of effluent treatment.

The filterable or settleable solids contain most of the phosphorus discharged from tanks, 50-85% (Cripps & Bergheim, 2000), but relatively little of the total effluent nitrogen, about 7-32% (Braaten, 1991; Heinen et al. 1996; Cripps & Bergheim, 2000). Most of the effluent nitrogen released (75–80%) is in the form of dissolved ammonia or nitrate when nitrification is promoted. The variability in the nutrient and organic material fractionation between dissolved and particulate matter is largely dependent on feed formulation and the opportunity for particulate matter to break apart, because the production of smaller particles increases the rate of nutrient and organic matter dissolution. Fecal matter, uneaten feed and feed fines can be rapidly broken down

into much finer and more soluble particles by water turbulence, fish motion, scouring along a tank/pipe bottom, and pumping. It is much more difficult to remove dissolved and fine particulate matter than larger particles. Therefore, culture tank designs and operating strategies that remove solids rapidly and with the least turbulence, mechanical shear, or opportunity for microbiological degradation are important to help the fish farm meet discharge limits (to be discussed in more detail below).

6.3 WASTE CHARACTERISTICS

A general mass balance on waste production from fish is shown in Fig. 6.1. Chen et al. (1993) summarized the waste production characteristics of the concentrated TSS coming from an RAS and compared it to domestic sludge characteristics (see Table 6.1). Compared with typical municipal sludge, aquacultural sludge has a relatively lower solid content and BOD_5 concentration. Total ammonia nitrogen (TAN) is fairly low for fresh aquacultural sludge, but can increase drastically if the sludge is left undisturbed for a period of time and mineralization occurs under anaerobic conditions. Aquaculture sludge has a higher nitrogen and phosphorous content than domestic sludge. The average value of total phosphorous (TP) is 1.3% of the dry solid mass, while the typical domestic sludge contains only 0.7%.

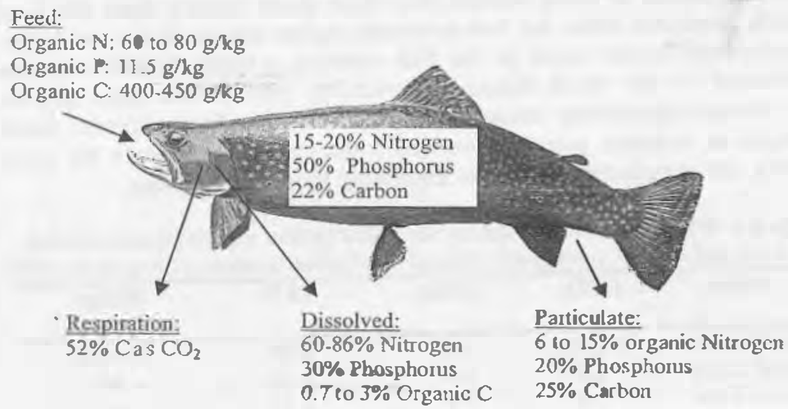


Figure 6.1 General mass balance on a feeding fish (Seabass). Note by book author: Carbon balance adds up to approximately 100% with regards to organic carbon diffusion. (D'orbcastel & Blancheton, 2006)

Table 6.1 Waste Production Characteristics of Sludge (Chen et al. 1993)

Parameter	Aquacultural Sludge Range	Mean	StDev	Domestic Sludge Range	Typical
Total Solids(%)	1.4–2.6	1.8	0.35	2.0–8.0	5.0
TVS (% of TS)	74.6–86.6	82.2	4.1	50–80	65
BOD_5 (mg/L)	1,590–3,870	2,760	210	2,000–30,000	6,000
TAN (mg/L)	6.8–25.6	18.3	6.1	100–800	400
TP (% of TS)	0.6–2.6	1.3	0.7	0.4–1.2	0.7
pH	6.0–7.2	6.7	0.4	5.0–8.0	6.0
Alkalinity	284–415	334	71	500–1,500	600
BOD_{20} (mg/L)	3,250–7,670	5,510	1,210	—	—

Focusing on solid waste, the physical properties of different fish manures can be dramatically different. Tilapia manure is distinctly different than, say; trout feces in that tilapia produce feces that are encapsulated in a mucous membrane resulting in sausage-like strands. The tilapia fecal strands will float to the surface about 30 minutes or so after being defecated due to gas bubbles being formed in the feces within the strand and then later sink again as gas bubbles diffuse through the membrane strands.

Carbohydrate makeup in tilapia feces will also have an impact on the fecal strand density. In general, higher carbohydrate diets and lower fat diets will result in fecal strands that float more readily than the fecal strands produced when the fish consume higher energy diets; the higher carbohydrate levels result in the fish creating a more definitive mucus membrane for the fecal strand. Digestibility also affects fecal strands, with lower digestibility resulting in more floating fecal strands. Small changes in nutrient composition can have dramatic impacts on water quality. Be extremely cautious in switching feed formulations.

Table 6.2 Waste Generation (kg/day per 1,000 kg live weight) from a Catfish RAS and Other Commercial Animal Production Systems (Chen et al. 1993)

Animal	BOD_5	Solids	TKN	Sludge Volume (L)
Catfish RAS	0.8–1.3	4.2	0.20	70-420
Beef Cattle	1.6	9.5	0.32	30
Dairy Cow	1.4	7.9	0.51	51
Poultry	3.4	14.0	0.74	37
Swine	3.1	8.9	0.51	76

Note: Phosphorus generation is approximately 1% of the feed fed.

Waste generation is covered in Chapter 4 but as a quick means of reference, Table 6.2 provides a comparison of the amount of waste generated by the major animal groups. Adjustments can be made where there is a better estimate of feeding rates for a particular aquaculture application.

6.4 Waste Management Overview

The two most common methods used to recycle solid wastes from aquaculture facilities are land application and composting. Most states have guidelines or regulations that govern land application of manure and other organic wastes to fertilize agricultural crops by limiting the land application rates and the amount of associated pathogens, heavy metals, and other contaminants. The land application rates are based on the nutrient content, soil type, and plant nutrient uptake characteristics to prevent runoff or groundwater contamination. Odor problems can also limit land application in developed areas. Finally, the transportation from the point of generation to the application site can be a major factor in the costs of sludge management, since aquaculture sludge, even when thickened, is mostly water.

Before either of these disposal methods can be used, the sludge and effluent wastewater first needs to be transferred to some form of storage system for both sludge thickening and flow equalization. Sludge thickening will reduce the hydraulic loading on subsequent processes. In addition, sludge flow rates and concentrations will vary during cleaning and other maintenance activities. Therefore, some form of flow equalization is needed to equalize the flow on subsequent treatment processes. Sludge storage structures include anaerobic lagoons, earthen ponds, and above and below ground tanks. Sludge thickening can be accomplished by settling basins, geotextile bags, belt filters, lime stabilization, wetlands, and sand beds. Often some form of coagulation and flocculation aid is added to enhance settling and in some cases sequester dissolved phosphorus.

6.5 Storage, Thickening, And Stabilization

Storage and Thickening

Thickening basins and separate storage structures are both utilized for storing aquacultural sludge. Thickening basins can be designed to accommodate the build-up of solids and provide temporary solids storage capacity. However, as sludge accumulates within these basins they will become less and less effective at solid-liquid separation due to

impingement on the proper settling hydraulics, solids flotation due gas bubble production from fermentation, and dissolution of nutrients and organic matter.

In many cases, the thickened sludge from thickening basins is transferred to larger sludge storage structures capable of holding months worth of captured and thickened solids. Sludge storage structures utilized include earthen ponds, above ground tanks, and below ground tanks. Earthen ponds are generally rectangular basins with inside slopes (horizontal:vertical) of 1.5:1 to 3:1. Depending on site geology and hydrology, earthen ponds can have liners of concrete, geomembrane, or clay. Earthen pond design will include the capacity for storage of precipitation as well as a method for removing solids. In the case where solids will be unloaded via pumping, the solids must be agitated to provide a uniform consistency. Pond agitation may be accomplished with hitch-type propeller agitators that are powered by tractors or by agitation pumps. Propeller agitators work well for large ponds, while chopper-agitator pumps work well for smaller ponds. Solids unloading may also be done with heavy equipment, in which case pond design should include ramp access (maximum slope of 8:1) and suitable load capacity in the unloading work area (NRCS, 1996).

Sludge may also be stored in tank structures, above and below ground. Storage tanks are primarily constructed of reinforced concrete, metal, and wood. Reinforced concrete tanks including walls, foundation, and floor slab-- may be cast-in-place or they may be constructed of pre-cast wall panels, bolted together, and set on a cast-in-place foundation and floor slab. Metal tanks are also widely used, with the majority being constructed of glass-fused steel panels that are bolted together. There are many manufactured, modular tanks commercially available in reinforced concrete and metal, as well as wood (NRCS, 1996).

Design of all structures, earthen or manufactured, should include considerations for internal and external hydrostatic pressure, flotation and drainage, live loads from equipment, and dead loads from covers and supports (NRCS, 1996).

SOLIDS THICKENING –SETTLING BASINS

Captured solids from solids removal processes tend to be dilute, having less than 2% total solids content. These solids may be concentrated or thickened in settling basins up to 5–10% total solids content (Bergheim et al. 1993). Solids thickening also significantly reduce disposal cost by reducing volume of sludge. For example, for 1000 kg dry weight of solids, the sludge volume at 1% solids is over 100 m^3, at 5% solids, 20 m^3, at 10% solids, 10 m^3, at 20% solids only 5.0 m^3 and at 30% solids only 3.3 m^3.

Thickening basins operate according to the same discrete particle settling principles previously described. However, because thickening basins receive water with elevated solids content and are concentrating these solids by permitting particle settling, the solids are also subject to compression settling. Compression settling develops when a compressed layer of particles forms at the basin bottom. The particles in this region begin to form a structure of particle-particle contact and the slurry is concentrated further (Crites and Tchbanoglous, 1998). In general, the overflow rate for sludge thickening basins should be approximately 1.0 $m^3/m^2/hr$ (3.2 $ft^3/ft^2/hr$) with hydraulic retention times of between 20 to 100 minutes (Bergheim et al. 1993; Bergheim and Cripps, 1998).

Off-line settling basins are designed for solids collection, thickening, and storage. They have been successfully used for the treatment of slurries from quiescent zone cleaning activities in raceway production, microscreen filter backwash water from recirculation systems and flow from system cleaning activities. The Conservation Funds Freshwater Institute has successfully applied the concept of settling basins to concentrating the backwash coming off the drum filters. Three off-line settling cones or thickening tanks are used to capture and store solids from the intermittent backwash of three drum filters. The solids-laden backwash flow is introduced intermittently into the top and center of each tank. At the top of each tank, the flow is introduced within a cylinder with an open bottom that is centered within the tank. The cylinder improves the hydraulics of the tank's radial flow by directing the water to first flow down (underneath the cylinder and towards the cone of the tank) and then up as it travels radially towards the effluent collection launder about the top circumference of the tank. These thickening tanks have performed well, capturing 97% of the solids discharged from the microscreen filter backwash flows. In addition, the three settling cones are plumb such that the three waste streams can be directed to a single cone or multiple cones (Ebeling & Summerfelt, 2002).

Figure 6.2 Off-line settling basins at the Conservation Funds Freshwater Institute used to concentrate backwash water from a microscreen filter.

The recommended design criteria by the Idaho Department of Environmental Quality (IDEQ, 1998) for off-line settling basins are an overflow rate of 40 m^3/day per m^2 (0.0015 ft^3/sec flow per ft^2) surface area and usually depth of 1.1 m (3.5 feet). Depth is not required for settling efficiency but is required to provide storage for solids; three and one half feet provides adequate storage for ponds in which solids are removed monthly.

SOLIDS THICKENING–COAGULATION/FLOCCULATION AIDS

One of the most commonly used methods for the removal of suspended solids in drinking water is the addition of coagulant and flocculation aids, such as alum, ferric chloride, and long chain polymers (AWWA, 1997). Coagulation/flocculation process is widely used in the drinking water treatment area as a means of attacking fine suspended (< 10 um) and colloidal particles (<1 um). It is normally implements as a four step process: 1) coagulation, 2) flocculation (floc formation), 3) sedimentation (gross floc removal) and 4) sand filtration (fine solids removal or polishing).

Coagulation is the process of decreasing or neutralizing the electric charge on suspended particles or zeta potential. Similar electric charges on small particles in water cause the particles to naturally repel each other and force the small, colloidal particles apart and keep them in suspension. This allows the van der Waals force of attraction to encourage initial aggregation of colloidal and fine suspended materials to form microfloc.

Flocculation is the process of bringing together the microfloc particles to form large agglomerations by physically mixing or through the binding action of flocculants, such as long chain polymers. Polymers or polyelectrolytes consist of simple monomers that are polymerized into high-molecular-weight substances (Metcalf and Eddy, Inc., 1991) with molecular weights varying from 10^4 to 10^6 Daltons. Polymers can vary in molecular weight, structure (linear versus branched), amount of charge, charge type and composition. With respect to charge, polymers can be cationic (positively charged), anionic (negatively charged) or nonionic (no charge). Polyelectrolytes act in two distinct ways: charge neutralization and bridging between particles. Because wastewater particles are negatively charged, low molecular weight cationic polyelectrolytes can act as a coagulant that neutralizes or reduces the negative charge on the particles, similar to the effect of alum or ferric chloride. This has the effect of drastically reducing the repulsive force between colloidal particles, which allows the van der Waals force of attraction to encourage initial aggregation of colloidal and fine suspended materials to form microfloc. These larger particles are then removed by sedimentation and/or filtration. A wide range of coagulation/flocculation aids have been employed, including alum, polyaluminum chloride (PAC's), polyaluminum sulfate (PAS) and anionic and cationic polymers and natural polymers such as chitosan. In the drinking water industry, the coagulation/flocculation process is used to produce potable water with turbidities below 1 NTU, water that is effectively free of suspended or colloidal solids.

Higher molecular weight polymers are generally used to promote bridging flocculation. The long chain polymers attach at a relatively few sites on the particles, leaving long loops and tails which stretch out into the surrounding water. In order for the bridging flocculants to work, the distance between the particles must be small enough for the loops and tails to connect two particles. The polymer molecule thus attaches itself to another particle forming a bridge. Flocculation is usually more effective the higher the molecular weight of the polymer. If too much polymer is used however, the entire particle surface can become coated with polymer, such that no sites are available to "bridge" with other particles.

Because the chemistry of wastewater has a significant effect on the performance of a polymer, the selection of a type of polymer for use as a coagulant/flocculation aid generally requires testing with the targeted waste stream and the final selection is often more of an "art" than a science. Hundreds of polymers are available from numerous manufactures with a wide variety of physical and chemical properties. Moreover, although the manufactures can often help in a general way,

the end user must often determine from all the various product lines, which is best for their particular application and waste stream.

Numerous substances have been used as coagulant and flocculation aids, including alum [$Al_2(SO_4)_3 \cdot 18H_2O$], ferric chloride [$FeCl_3 \cdot 6H_2O$], ferric sulfate [$Fe_2(SO_4)_3$], ferrous sulfate [$FeSO_4 \cdot 7H_2O$] and lime [$Ca(OH)_2$] (Metcalf and Eddy 1991).

Alum

Aluminum sulfate or alum [$(Al_2(SO_4))_3 \cdot 18\ H_2O$] has long been used in the wastewater treatment industry as an inorganic coagulant due to its reliable performance, widespread availability, low cost, and ease of handling. The overall effectiveness of alum is dependent on the water chemistry, especially pH and alkalinity. Alum works best in the pH range of 5.5-8.0 which is compatible with most RAS applications. Alum consumes 0.45 mg/L as $CaCO_3$ for every 1.0 mg/L of Alum dosed employed (Metcalf and Eddy, 1991), a demand that should not present a problem for RAS applications that are usually a managed to maintain alkalinities above 150 mg/L as $CaCO_3$. When alum is added to water, the following reaction takes place:

$$Al_2(SO_4)_3 \cdot 18\ H_2O + 3\ Ca(HCO_3)_2 \Leftrightarrow 3\ CaSO_4 + 2\ Al(OH)_3 + 6\ CO_2 + 18\ H_2O \qquad (6.1)$$

The insoluble aluminum hydroxide, $Al(OH)_3$, is formed as a gelatinous floc that sweeps out fine suspended and colloidal solids material. In addition, when alum is added to water, the aluminum salts (Al^{+3}) reacts quickly with any dissolved phosphate ions to produce aluminum phosphate which precipitates with the aluminum hydroxide:

$$Al^{+3} + PO_4^{-3} \Leftrightarrow AlPO_4 \qquad (6.2)$$

The above equations are the simplest forms of the reaction (Metcalf and Eddy, 1991). Due to the many other competing reactions, the effects of alkalinity, pH, trace elements, and other compounds in the wastewater, the actual chemical dosage required to remove a given quantity of phosphorus is usually established on the basis of bench-scale test or sometimes pilot-scale tests.

More recently, several manufacturers have begun to market lanthanum chloride as a safe, non-toxic phosphate remover. The lanthanum chloride works by absorbing phosphate ions while releasing chloride ions acting like an organic ion exchanger which is highly selective towards phosphate ions. More importantly, it has no impact on pH or alkalinity. When lanthanum chloride as added to water, the following reaction takes place extremely rapidly,

$$LaCl_3 + PO_4^{-3} \Leftrightarrow LaPO_4 + 3\ Cl^- \qquad (6.3)$$

The insoluble lanthanum phosphate, $LaPO_4$, is formed as a small particle that is removed by settling or filtration.

Polymers and Poly-Aluminum Chlorides (PACs)

Polymers or polyelectrolytes consist of simple monomers that are polymerized into high-molecular-weight substances (Metcalf and Eddy, 1991). Polymers can vary in molecular weight, structure (linear versus branched), amount of charge, charge type, and composition. The intensity of the charge depends upon the degree of ionization of the functional groups, the degree of copolymerization, and/or the amount of substituted groups in the polymer structure (Wakeman and Tarleton, 1999). With respect to charge, organic polymers can be cationic (positively charged), anionic (negatively charged), or nonionic (no charge). Polymers in solution generally exhibit low diffusion rates and raised viscosities, thus it is necessary to mechanically disperse the stock solution of polymer into the water being treated. This is accomplished with short, vigorous mixing to maximize dispersion, but not so vigorous as to degrade the polymer or the flocs as they form (Wakeman and Tarleton, 1999).

Polymers, in contrast to alum, are not pH dependent and have no known effect on pH or alkalinity. They can increase the rate of flocculation and produce larger and denser flocs that settle faster and help to improve filtration. They are, however, more expensive and are specific to certain water quality characteristics. Because the chemistry of wastewater has a significant effect on the performance of a polymer, the selection of a type of polymer for use as a coagulant/flocculation aid generally requires testing with the targeted waste stream Hundreds of polymers are available from numerous manufacturers with a wide variety of physical and chemical properties.

There has been some concern about the toxicity of polymers to the environment. However, in addressing these concerns, EPA has determined that "upon closer review of the matter, it appears that this concern has been raised due to anecdotal suggestions, rather than documented evidence of actual discharge events causing toxic effects. To date, EPA has not identified any documented cases where the use of a polymer to treat C&D storm water discharges caused an adverse effect in the receiving waters."

Chitosan is an organic cationic polymer that has also been used as a flocculating agent in the treatment of wastewater and food industry. Chitosan is a carbohydrate biopolymer derived from the polysaccharide chitin, the structural component of the exoskeletons of crustaceans.

Chitosan is a biodegradable, non-toxic, linear cationic polymer of high molecular weight. The ability of chitosan to biodegrade in the environment makes it a sustainable, environmentally viable alternative to chemical treatment.

The effectiveness of commercial coagulation-flocculation polymers and/or alum for removing both suspended solids and phosphorus from aquaculture wastewater flows has been intensively researched by the author. Replicated jar test studies have determined:

- the optimum alum and ferric chloride dosages (when used separately) and flocculation conditions (e.g., mixing speed and time) required to reduce suspended solids and total phosphorus concentrations in the supernatant overflow from gravity thickening tanks (Ebeling et al., 2003);
- the most advantageous alum or ferric chloride concentrations and conditions for coagulation, flocculation, and settling of suspended solids and phosphorus found in the backwash flow discharged from a microscreen drum filter (Ebeling et al., 2004);
- the most suitable polymer type and the appropriate polymer dose, mixing speed and time, and flocculation conditions required to maximize suspended solids and particulate phosphorus removal from the backwash flow discharged from a microscreen drum filter (Ebeling et al., 2005);
- the optimum combination of alum and polymer concentrations and mixing and flocculation conditions to remove suspended solids and phosphorus from the backwash flow discharged from a microscreen drum filter (Rishel and Ebeling, 2006).

The optimum alum dosage was tested by one of the authors using microscreen backwash water under a set of standard mixing and flocculation settings (Ebeling et al., 2003). Optimal turbidity removal was achieved with a 60 mg/L dosage of alum, reducing the average initial TSS values of approximately 320 mg/L to approximately 10 mg/L. In addition, the orthophosphate removal efficiencies for alum and ferric chloride were greater than 90% at a dosage of 60 mg/L with final concentrations of Reactive Phosphorus (RP) approaching 0.15 mg/L-P. Flocculation and mixing intensity and duration played only a minor role in the removal efficiencies for both orthophosphates and suspended solids. Both coagulation-flocculation aids also exhibit excellent settling characteristics, with the majority of the floc quickly settling out in the first 5 minutes.

As part of a research project conducted by one of the authors, three commercial sources of polymers for the wastewater industry were contacted and samples obtained of recommended polymers for aquaculture wastewater (Rishel & Ebeling, 2006). The companies were Ciba Specialty Chemicals Corporation, http://www.cibasc.com; Cytec Industries Inc. http://www.cytec.com; and Hychem, Inc., http://www.hychem.com. Eighteen different polymers were obtained with a wide range of chemical families, charge densities, and molecular weights. The coagulation-flocculation tests of the polymers were again carried out following the standard practice for coagulation-flocculation testing of wastewater (ASTM, 1995).

A series of jar tests were used to initially screen each of the eighteen polymers and estimate optimal dosage and percent removal of total suspended solids and reactive phosphorus. Based on these results, six polymers were chosen for further study. Three of the polymers had a very high degree of cationic charge; two have a high degree of cationic charge, and one has a low degree of cationic charge. In addition, three have a very low molecular weight, one has a high molecular weight, and two have a very high molecular weight. No anionic charged polymers were chosen due to their low overall performance. Magnafloc LT 7991, 7992, and 7995 have a very high degree of cationic charge and a low molecular weight so should operate very similarly to coagulants like alum by adsorption-charge neutralization of particles. Hyperfloc CE 854 and CE 1950 have both a high degree of cationic charge and a high molecular weight and should provide both charge neutralization and bridging between particles. Magnafloc LT 22S with a very low degree of cationic charge and a high molecular weight should work primarily by bridging between particles.

Although a wide range of polymers were tested, the results show excellent removal efficiencies for all of them. Total suspended solids removal was close to 99%, with final TSS values ranging from as low as 10 to 17 mg/l. Although not intended to be used for reactive phosphorus removal, reactive phosphorus was reduced by 92 to 95% by removing most of the TSS in the wastewater to approximately 1 mg/l – P. Dosage requirements were uniform, requiring between 15 and 20 mg/l of polymer.

A series of screening tests were then conducted using both alum as a coagulation aid and the initial 18 polymers. Based on preliminary tests with alum on the microscreen effluent discharge, a dosage of from 50 to 60 mg/L was found to yield the best overall removal of TSS and Reactive Phosphorus and thus was used as a guide for all the screening and evaluation tests. The polymers screened were of assorted chemical families, electric charge, molecular weight, and all but three had maximum dosage limits set for potable water by the National Sanitation Foundation. These limits helped determine the dosage range to be tested. The main purpose of the screening was to examine the performance of each alum / polymer combination at several different polymer

concentrations, by looking specifically at their ability to remove suspended solids and phosphorus from the treated water. The effectiveness and efficiency of each combination was determined by comparing the initial water quality and the treated water quality. In addition, a control was carried through the Jar Test procedure. At the end of the screening, six polymers and optimal dosage rates were picked for further evaluation. They were selected primarily based on their outstanding performance, but also intentionally selected to have varying chemical composition/structure. For example, three of the polymers chosen exhibited a cationic charge and three an anionic charge. In addition, the degree of electric charge varied from low, medium, high to very high. The same was seen for molecular weights with one very low, two high and two very high.

Table 6.3. Initial Sample and Treated Sample Total Suspended Solids Concentrations and Removal Efficiencies at the Optimal Dosage Levels for Selected Polymers in Combination with 50 mg/L alum (Rishel & Ebeling, 2006)

		Total Suspended Solids (mg/L)		
Polymer / Chemical Family	Optimum Dosage	Raw sample (Mean.)	Treated sample (Mean.)	Percent Removal
LT27	0.8 mg/L	**557**	**7**	**99%**
Copolymer of sodium acrylate and acrylamide		96	1.7	
LT 7995	6 mg/L	**859**	**10**	**99%**
Organic cationic polyelectrolyte		583	1.3	
E 38	3 mg/L	**1566**	**20**	**98%**
Anionic polyacrylamide emulsion		1469	5.5	
A-120	0.8 mg/L	**654**	**7**	**99%**
Anionic polyacrylamide		181	4	
CE 834	5 mg/L	**719**	**4**	**99%**
Cationic polyacrylamide		193	1	
CE 1950	5 mg/L	**958**	**10**	**99%**
Cationic polyacrylamide		200	6	

Table 6.3 and 6.4 summarize the polymers tested and the removal efficiencies for TSS and reactive phosphorus. Although a wide range of polymers were used, i.e. chemical families, charge density, molecular weight, results showed excellent removal efficiencies for all of them. Using a combination of alum/polymer, the effluent Total Suspended Solids removal rate was close to 99%, with final TSS values ranging from as low as 4 to 20 mg/L. Reactive phosphorus was reduced by 92 to 99% to as low as 0.16 mg/L-P. Finally, Total Phosphorus was also significantly reduced (98%) with treated effluent concentrations from 0.9 to 3.0 mg/L-P.

Table 6.4. Initial Sample and Treated Sample Reactive Phosphorus Concentrations and Removal Efficiencies at the Optimal Dosage Levels for Selected Polymers in Combination with 50 mg/L alum (Rishel & Ebeling, 2006)

		Reactive Phosphorus (mg/L P)		
Polymer / Chemical Family	Optimum Dosage	Raw sample (Mean.)	Treated sample (Mean.)	Percent Removal
LT27	0.8 mg/L	**10**	**0.17**	**98%**
Copolymer of sodium acrylate and acrylamide		2.9	0.04	
LT 7995	6 mg/L	**17**	**0.26**	**98%**
Organic cationic polyelectrolyte		15.6	0.06	
E 38	3 mg/L	**34.8**	**0.57**	**98%**
Anionic polyacrylamide emulsion		32.2	0.41	
A-120	0.8 mg/L	**11.4**	**0.16**	**99%**
Anionic polyacrylamide		4.5	0.02	
CE 834	5 mg/L	**13.7**	**0.27**	**98%**
Cationic polyacrylamide		7.3	0.22	
CE 1950	5 mg/L	**17.1**	**0.35**	**98%**
Cationic polyacrylamide		5.1	0.20	

Although the suspended solids and reactive phosphorous in the discharged effluent was the primary focus of this research, several other parameters were evaluated in a series of separate tests at the optimum polymer dosage, Table 6.5. These included ammonia-nitrogen, nitrite-

nitrogen, nitrate-nitrogen, total nitrogen, $cBOD_5$, and COD. Although not intended for nitrogen removal, TAN, nitrite-nitrogen, nitrate-nitrogen, and total nitrogen in the effluent were reduced on average by 64%, 50%, 68%, and 87% respectively. Removal rates for both $cBOD_5$ and COD were also significant, with an average value of 97.3% and 96.4%.

Table 6.5. Impacts of Alum as Coagulation Aid and Polymer as a Flocculation Aid on Selected Additional Water Quality Parameters; mean and standard deviation of triplicate samples (Rishel & Ebeling, 2006)

	TAN (mg/L N)	NO_2-N (mg/L N)	NO_3-N (mg/L N)	TN (mg/L N)	$CBOD_5$ (mg/L)	COD (mg/L)
Initial Sample	0.75	0.430	10.8	34	437.7	719
LT 27	0.32	0.218	3.6	4.8	17.8	36
LT 7995	0.28	0.216	3.7	4.4	8.1	21
E 38	0.24	0.224	3.7	4.7	12.0	27
A-120	0.36	0.222	3.6	4.3	17.7	29
CE 834	0.19	0.191	2.7	3.5	7.7	20
CE 1950	0.24	0.219	3.6	4.5	8.9	21

Solids Thickening – GeoTextile Bags

One promising new technology for dewatering aquaculture solid waste is the use of geotextile bags. Geotextile bags are porous sealed tubular containers constructed of a woven polyethylene material and have demonstrated the ability to dewater animal wastes, municipal wastewater sludge; hazardous wastes, industrial by-products, and dredge spoil. It has been demonstrated that geotextile bags can dewater wastes to over 10% solids in less than a week, and can achieve final solids content over 30% over several months. Geotextile bags are cost effective, site-specific, and mobile, require little maintenance, and can be manufactured for high containment volumes. They are currently available from several aquaculture and agricultural supply companies.

Research trials conducted by the author with small lab-scale Geotube® geotextile bags (Ebeling & Rishel, 2006) showed that when combined with a polymer flocculation aid, they demonstrated excellent potential to dewater solid wastes generated by a rotating microscreen filter in an intensive recirculating aquaculture system. The small scale demonstration Geotube® geotextile bags combined with a polymer flocculation aid removed an average 93% of the total suspended solids in a one month period. In addition, substantial removal of reactive phosphorus, and some removal of total nitrogen and BOD was seen. One

potential problem with geotextile bags is the leaching of ammonia-nitrogen, which may require addition treatment of the leachate before discharge.

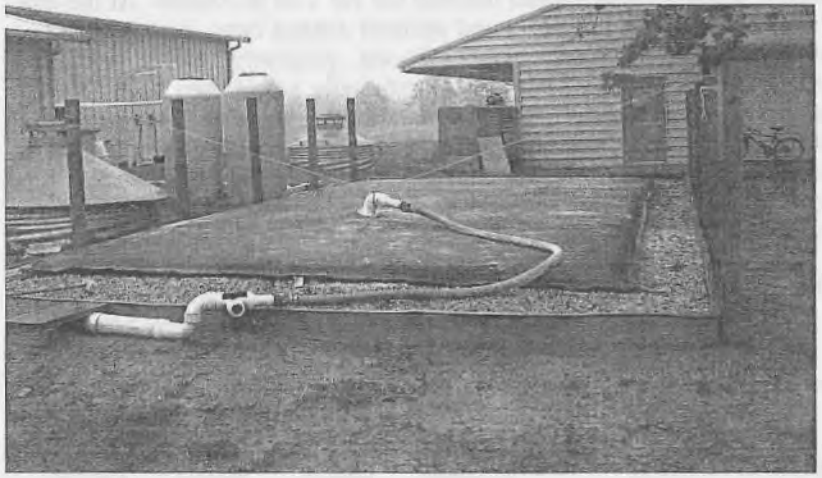


Figure 6.4. A GeoTextile Bag used to treat waste from an intensive tilapia system at North Carolina State University, Fish Barn.

North Carolina State University Fish Barn

Geotextile bags have been used at several aquaculture research programs, although there has been very little data presented or published by these researchers. One of the first to use this process for waste treatment was Dr. Tom Losordo at the North Carolina State University Fish Barn, where Geotextile bags have been used to treat tilapia wastes from several microscreen filters for ten years. To date, one research abstract has been presented at the 2006 WAS meeting in Florence, Italy on the systems.. In the NCSU design approach, waste water generated from the backwash cycle of a microscreen filter (40 micron screen) is first collected in a small sump and then pumped at 2.3 m^3/hr (10 gpm) into a geotextile bag with a 400 micron pore size. An organic polymer (Hychem Hyperfloc CE 1950 or CE 834 Polymer) is injected into this flow stream with a standard diaphragm pump at an unspecified rate and mixing occurs in a serpentine path of PVC elbows. The Geotextile bag rests upon a gravel bed and the soluble effluent drains through the bag and is collected a second sump. Limited research data has been reported but the WAS abstract reported that the influent suspended solids concentration averaged 1200 mg/L and the average effluent concentration was 44 mg/L, a reduction of over 96%. In addition,

denitrification processes within the geotextile bag resulted in a passive reduction of the nitrate-nitrogen from an average of 144 mg-N/L to 74 mg-N/L or 50%. In addition, Total Phosphorus was reduced by an average of 34%, and COD by 87%.

Mote Marine Laboratory

The Mote Marine Laboratory in Florida has used geotextile bags for treating wastes from a demonstration intensive marine shrimp system and more recently for a sturgeon demonstration facility. There have been no published reports on their design or performance. Mote Marine Lab is also working on a project entitled the Design and Evaluation of Marine Fish and Live Feed Recirculating Systems for Inland Aquaculture with the State of Florida's Division of Aquaculture, Department of Agriculture and Consumer Service (Main and Losordo, 2009). The goal of this project was to design, construct and evaluate innovative recirculating systems to advance the development of a sustainable marine aquaculture industry in Florida. As part of this project a waste treatment systems had been designed consisting of a waste sump to accumulate solids, a containment system for solids or geotube bag, a polymer tank to facilitate solids settlement in the geotube, and a sterilization system that includes solids and biofiltration, UV sterilization and ozone to cleanse the water prior to pumping it back to the fish culture system. No other details are available.

Reymann Memorial Farm, Wardensville, WV

A small demonstration project was conducted at the Reymann Memorial Farm, Wardensville, WV by Dr. Karen Buzby and Jennifer Hendricks, investigators in the Environmental Engineering Department at West Virginia University. This project was to show if geotextile bags could be used to treat routine wastes collected from the quiescent zones in fish raceways. The solid waste , which had settled at the end of the trout raceway, flowed by gravity first to a sludge sump, where it was then pumped into a geotextile bags. In this demonstration, a total volume of approximately 5.7 m^3 (1500 gallons) was removed from the quiescent zones of the trout raceways

at least three times each week. In an effort to maximize recovery of solid waste, a flocculant aid (Hyperfloc® CP625, Hychem Inc.) was added as the water was pumped into a 4.5 x 7.5 m (15 x 25ft) geotextile bag.

Drainage pipes in a gravel pad directly under the geotextile bag allowed the permeate to flow to a central collection point, where it was discharged to a pond. Influent to and effluent from the geotextile bag were analyzed for total suspended solids, BOD_5 and particle size several weeks after installation. Initial results showed that the addition of 20 mg/L DADMAC type polymer with high molecular weight and viscosity removed 99% of Total Settleable Solids (TSS) and 87% of the BOD. Effluent nutrient concentrations increased over influent concentrations in samples taken 3 months after operation. NH_3-N concentrations increased from 2.6 to 3.1 mg/L, NO_3-N concentrations increased from 0.3 to 1.5 mg/L and PO_4 effluent concentrations increased from 6.4 to 8.9 mg/L. NO_2-N concentrations increased from 0.015 mg/L in the influent to 0.39 mg/L in the effluent. This indicated that decomposition was occurring within the geotextile bag and some of the nutrients were being released into the supernatant flow.

The Conservation Funds Freshwater Institute

The Freshwater Institute has conducted tests with small scale geotextile bags and have published the results in Aquaculture Engineering (Sharrar, et al, 2009). In their design, the microscreen backwash discharged from several intensive recirculating aquaculture systems was treated and dewatered using lab scale geotextile bag filters.

Three chemical coagulation aids (aluminum sulfate (alum), ferric chloride, and calcium hydroxide (hydrated lime), were tested in combination with a long-chain polymer flocculation aid (Hychem CE 1950 at 25 mg/L) to determine the most cost effective and efficient treatment combination. Three different coagulants were tested to determine if coagulant choice impacts nutrient and carbonaceous biochemical oxygen demand ($cBOD_5$) leaching into the filtrate and the final composition of the bag-captured biosolids at the end of each period. Results from replicated geotextile bag filter tests indicated that when alum, ferric chloride, and hydrated lime (plus a polymer) were amended to a backwash flow, both suspended solids capture and solids thickening were improved; e.g., total suspended solids removal rates of 95.8, 95.1, and 96.0%, respectively, were achieved along with final dewatered filter cake percent solids concentrations of 22.1, 19.3, and 20.9%, respectively.

Alum, ferric chloride, and hydrated lime (plus a polymer) amended geotextile bags were not as effective in chemical oxygen demand (COD) and $cBOD_5$ removal, resulting in removal rates of 69.6, 67.2, and 35.3%, respectively, and 56.6, 9.3, and 47.4%, respectively. Total nitrogen removal applying alum, ferric chloride, and lime were also less than effective, resulting in removal rates of 39.1, 46.7, and 8.9%, respectively. Filtrate total nitrogen concentrations were primarily in the inorganic form (total ammonia nitrogen) suggesting mineralization of ammonia as solids were stored within geotextile bags under anaerobic conditions. Alum, ferric chloride, and lime amended bags were moderately efficient at total phosphorus removal, resulting in removal rates of 67.6, 47.0, and 77.3%, respectively. Alum was identified as the most cost effective chemical for coagulation, but hydrated lime was the most effective at dissolved phosphorus precipitation and removal

Table 6.6. Impacts of Alum as Coagulation Aid and Polymer as a Flocculation Aid (Sharrar, et al, 2009)

	Bag Influent	Bag Effluent	% Removal
TSS (mg/L)	1875 ± 811	98±25	93.0 ± 3
Total Phosphorus (mg/L)	40.6 ± 16	12.7±4.1	65 ± 12
Dissolved Reactive P (mg/L)	1.1 ± 0.7	10.8 ± 3.2	-1145 ± 574
Total Nitrogen (mg/L)	63.8 ± 25	37.9 ± 12	32± 24
TAN (mg/L)	1.7 ± 0.6	28.1 ± 9.9	-1587 ± 490
cBOD5 (mg/L)	517 ± 241	309± 80	47 ± 15

SOLIDS THICKENING – BELT FILTERS

One of the newest technologies to manage aquaculture effluent waste streams is the Belt Filter System, specifically designed to thicken sludge from the backwash water of a microscreen filter. When used in conjunction with coagulation / flocculation aids, significant reductions of suspended solids and soluble phosphorus are possible. By eliminating the need for settling tanks or ponds, the leaching of nutrients (phosphorus, nitrogen) is minimized and the dewatered sludge is in a form for easy transport, storage, or disposal. Alum is often used as the primary coagulation aid because it is readily availability in dry form and its ease of storage and mixing. A commercially available polymer is used as the flocculation aid. The coagulation/flocculation pretreatment system treated the waste discharge stream from a microscreen filter used as final treatment of discharge water from several large-scale recirculating aquaculture production systems at the Conservation Fund Freshwater Institute.

The Hydrotech belt filter (Figure 6.5) was designed to thicken sludge from the backwash water of a microscreen filter when used in combination with a coagulation/flocculation mixing system. This particular system is available from Water Management Technologies, Inc. (Baton Rouge, LA, http://www.w-m-t.com) and consisted of two parts, a mixing/flocculation tank and an inclined belt filter. The mixing tank is separated into four chambers, the first and last chambers have variable speed mixing impellers for slow mixing, and the smaller, intermediate chamber had a fixed, high speed impeller for polymer mixing. As the waste stream enters the first chamber, alum is injected with a variable speed, peristaltic pump from a reservoir at a predetermined dosing rate, i.e. mg of alum per liter of wastewater flow. The variable speed impeller mixes the alum with the wastewater stream and begins the coagulation process.

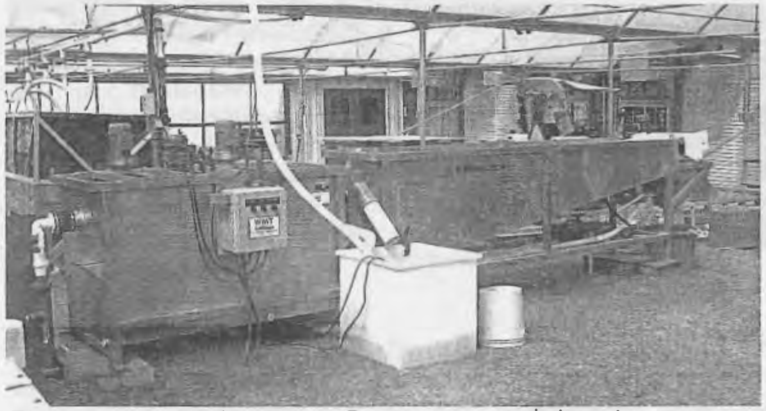


Figure 6.5 The Hydrotech belt filter system, consisting of a coagulation/flocculation tank and an inclined belt filter.

The fine particles in the wastewater stream are charge neutralized and begin to aggregate into small floc. The wastewater stream then flows over a weir into the smaller chamber, where polymer is injected at the surface, again using a variable speed peristaltic pump from a reservoir. A high rpm, fixed speed impeller mechanically mixes the polymer into the wastewater stream with a short, vigorous mixing to maximize dispersion of the polymer and force the wastewater stream down and into the third chamber. There the polymer begins the process of aggregation of the small particles and floc. Finally, the wastewater stream flows over a weir into the fourth chamber, where a variable speed impeller helps flocculate

the floc particles into larger floc and maintain them in suspension. From here, the waste stream consisting of large floc particles and relatively clear filtrate flows into the belt filter header box through a 10 cm pipe.

The continuous belt of the inclined belt filter (Fig. 6.5 and Fig. 6.7) is made of polyester cloth with a mesh size of approximately 120 microns and an angled inclination slope of 10 degrees. As the waste stream flows onto the belt and the filtrate passed through into the lower sump, the belt slowly becomes blocked by sludge and the headloss across the belt filter increases until a level sensor triggered the motor, which starts the endless band. The inclined belt filter then gently lifts the floc out of the water and transports it to the end of the belt where it is scraped off the belt by means of a firm rubber scraper (Fig. 6.7). A wash water jet spray system then cleans the belt before it rotates back to the inlet end. The wash water can be obtained either from a separate clean water source or from the filtrate water in the lower sump. The belt wash water is typically routed back to the head of the microscreen for further processing. As the belt is self cleaning, maintenance is kept to a minimum. In this particular application at the Conservation Funds Freshwater Institute, the clarified, treated wastewater stream flowed into a pump sump and to an aerobic wastewater treatment pond. The concentrated solids sludge were mixed with straw and as needed transported to a compost facility on site. If for some reason the belt filter was unable to process all of the influent flow, a by-pass weir diverted the untreated waste stream back to the head of the microscreen filter.

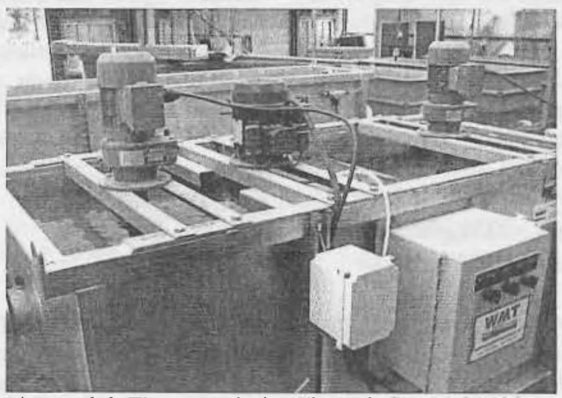


Figure 6.6. The coagulation/flocculation tank with two variable speed mixers and one fixed speed polymer mixer and mixers control panel.

Figure 6.7. Belt Filter Showing Inlet Weir box, Floc, Scraper Bar and Solids Sump.

During evaluation tests (Ebeling et al., 2006), alum used alone as a coagulant aid was nominally efficient in removing solids (83%), but was very efficient in sequestering reactive phosphorus (96%), with effluent concentrations less than 0.07 mg/L-P at 100 mg/L alum. A cationic polymer used alone and at relatively low dosages (15 mg/L) was very efficient in removing suspended solids, with a removal rate averaging 96% and an effluent TSS concentration of less than 30 mg/L. At the optimum dosage of alum and polymer, the Hydrotech Belt Filter System increased the dry matter content of the sludge to approximately 12.6 % solids, and reduced both the suspended solids and soluble phosphorus concentration of the effluent by 95% and 80% respectively. In addition, significant reductions in total phosphorus, total nitrogen, $cBOD_5$, and COD were seen (Table 6.7).

Table 6.7. Impacts of the Hydrotech belt filter system on water quality with alum as coagulation aid and polymer (Hychem, CE 1950) as a flocculation aid (Ebeling et al., 2006)

Alum/Polymer Dosage		pH	TSS (mg/L) Mean:	RP (mg/L-P) Mean:
0 mg/L / 0 mg/L (11)	Influent	7.37	1128	1.59
	Effluent	7.39	195	0.95
	% Removal		**81%**	**38%**
12.5 mg/L / 2.5 mg/L (11)	Influent	7.23	1120	1.81
	Effluent	7.26	110	0.67
	% Removal		**90%**	**59%**
12.5 mg/L / 5 mg/L (11)	Influent	7.26	1600	1.97
	Effluent	7.22	81	0.82
	% Removal		**94%**	**55%**
25 mg/L / 2.5 mg/L (18)	Influent	7.34	753	1.28
	Effluent	7.27	65	0.45
	% Removal		**91%**	**57%**
25 mg/L / 5 mg/L (3)	Influent	7.30	753	1.39
	Effluent	7.13	53	0.42
	% Removal		**93%**	**65%**
50 mg/L / 2.5 mg/L (13)	Influent	7.38	646	0.88
	Effluent	7.14	34	0.18
	% Removal		**95%**	**80%**

Solids Thickening – Reed drying beds

Depending on the location and local regulations, an aquaculture facility's only available options for sludge disposal may be limited and costly. However, if transportation costs make sludge disposal on cropland uneconomical, disposing of the sludge on-site within created wetlands may be an attractive alternative. A constructed reed drying bed can provide on-site treatment of a concentrated solids discharge with an uncomplicated, low-maintenance, plant-based system that could reduce solids disposal costs.

Reed drying beds are vertical-flow wetland (VFW) systems that have been used over the past 20 years to treat thickened sludge (1–7% solids) produced in the clarifier underflow at wastewater treatment plants. When used for municipal treatment, these wetlands are loaded with 7 to 10 cm

(2.76–3.94 inches) of 2% solids approximately once every 7–21 days (about 30–60 kg/m^2/yr). Although, the specific application and the nature of the wastewater will have an impact on any sustained level of infiltration.

Reed drying beds are also beginning to be used in aquaculture (Summerfelt et al. 1999). Summerfelt applied trout wastewater (7,800 mg/L total suspended solids) 6 times per day at a rate of 1.35 cm/day (30 kg dry solids per m^2 per year). During operation, a series of vegetated beds receives sequential batch applications of sludge. The sequential batch applications are such that the more recently flooded VFW cells are dewatering, while beds with older sludge applications are drying. Intervals between repeated sludge applications allow for dewatering and drying. Plants facilitate dewatering by conducting water along their stem and root paths through previous sludge layers and by removing water through evapo-transpiration. The plants also increase biological stabilization of the solids by transporting oxygen to their root zones. Reed bed treatment systems have been reported to have a useful lifetime of up to 10 years.

Solids Thickening – Rapid Sand Beds

The above section on reed beds is based upon the inherent hydraulic conductivity of the underlying sand bed. In the simplest form, one could design a dewatering bed that had no surface plants. In this case, you must design based upon the bed's hydraulic conductivity, or the coefficient of permeability, which is the standard measure of how quickly water flows through a soil (sand) column. Saturated hydraulic conductivity can be determined using Darcy's law (Ritzema, 1994). Sand beds may also incorporate marsh plants, such as phragmites, to sustain the hydraulic conductivity as described. In addition, removing such plants on a scheduled basis will also serve beneficially to remove nutrients from the wastewater (Ritzema, 1994; Kadlec and Knight 1996):

- Nitrogen ~225kg/ha/yr
- Phosphorus ~35kg/ha/yr

Sand bed drainage systems enhanced with plant cover crops have been used for a variety of applications including at least one commercial fish farm in Amherst, MA (Bioshelters, Mr. John Reid President, a 200,000 kg/year tilapia farm) and have been described in detail by others (Ritzema, 1994; Sanford et al. 1995; Kadlec and Knight, 1996).

Palacios and Timmons (2001) developed predictive equations of infiltration including the effects of biosolids accumulation (the major resistance to infiltration). These equations are as follows:

$$\frac{Q}{A} = -K_s \frac{\Delta H}{L} \tag{6.1^a}$$

$$\frac{\Delta H}{L} = \frac{Y/2}{N Y \beta} \tag{6.2}$$

$$L = \frac{C_{TSS}}{16} \beta D \tag{6.3}$$

where, K_S = 2.44 m/d and β is 0.05 m/m (based upon using tilapia wastewater with a TSS of 16 g/L).

These equations were developed from fish manure obtained from a commercial tilapia farm using RAS and employing approximately a 10% system volume exchange rate per day. The farm fed a 42% protein 12% fat feed; feed conversions at the farm were approximately 1 to 1. The TS concentration of the wastewater applied in these experiments was 2%, of which approximately 16 g/L was TSS (80% of the 2% was organic solids). Total manure depth (L) is the summation of solid layers accumulating from repetitive applications. The estimate of total L using Eq. 6.3 is used in Eq. 6.1 along with the K_S value of 2.44 m/day to predict infiltration rate. Note that the K_S value was specifically developed from tilapia feces and thus may be different for other fish manures. The filter medium is not expected to have any significant impact on estimating infiltration rates, since the K_S for the sand type medium is >> than the K_S for the manure. As a conservative rule of thumb, assume a design infiltration rate after repetitive applications of 3.6 cm/day.

"Rule of Thumb"
Sand beds will have an infiltration rate of 3.6 cm (1.4 inches) per day.

Managing Sand Beds

Treatment wetlands should be divided into smaller beds that are easier to manage. Operate these beds in parallel to better handle variable discharge volumes by flooding only the necessary area, or to ensure that treatment is not completely interrupted if one of the beds needs servicing. Beds also could be operated in series to achieve more complete treatment of the wastewater.

[a] Symbols are defined in List of Symbols at the end of the Chapter.

Using Eqs. 6.1 and 6.3 to predict infiltration rate probably provides a conservative estimate, since the rate equation is for sand beds without any surface vegetation. Use of phragmites or other emergent type monocot herbs would provide some continual disruption of the sand column due to root and shoot growth and probably to the manure surface layer as well. All disturbances cause large increases in infiltration. Consideration should also be given as to how one can restore hydraulic conductivity after repeated applications of wastewater. Simply removing the accumulated biomass and skimming a top layer of the sand materials will restore much of the sand bed's initial conductivity.

Solids Stabilization – Lime Addition

Thickened sludge in the bottom of thickening basins may be further conditioned by adding lime. Lime addition is an effective method to kill sludge pathogens, Table 6.8, reduce odor problems, and improve solids thickening. It is recommended that 15–20 g (0.125–0.167 lbs) of unslaked lime (CaO) per gallon of sludge (10% solids content) be used to achieve a pH of 12 for lime stabilization, Fig. 6.8. In addition to stabilizing the sludge and improving its settling properties, lime stabilization also increases the removal of phosphorus (Bergheim and Cripps, 1998, Bergheim et al. 1998).

Table 6.8 Sludge Lime Treatment of Aeromonas Salmonicida and IPN-virus Sludge at Different pH Values and Temperatures of 15 to 20°C (from Bergheim et al. 1998)

Time	*Aeromonas salmonicida*				IPN-virus	
	pH	$\frac{CFU}{mL}$	pH	$\frac{CFU}{mL}$	pH	$TCID_{50}$ mL^{-1}
0	11.0	3.0×10^6	12.0	3.0×10^6	11.9	$10^{8.3}$
1 hr	11.0	2.1×10^3	12.2	$<1 \times 10^3$	11.9	$10^{7.6}$
1 day	—	—	—	—	11.6	$10^{6.1}$
3 days	10.5	$<1 \times 10^3$	11.4	$<1 \times 10^3$	—	—
7 days	—	—	—	—	11.4	$10^{5.1}$
10 days	10.6	$<1 \times 10^3$	11.4	$<1 \times 10^3$	—	—

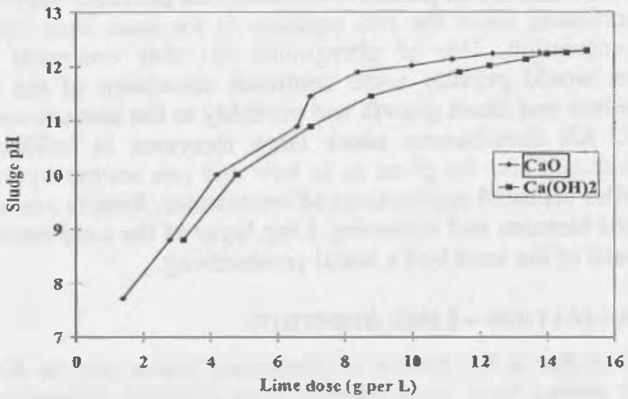


Figure 6.8 Effect of lime dose on aquacultural sludge pH for two types of stabilization amendments, CaO and $Ca(OH)_2$. Sludge dry matter content was 11.1% (from Bergheim et al. 1998).

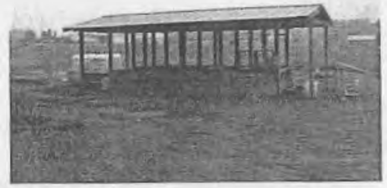
6.6 UTILIZATION/DISPOSAL

LAND APPLICATION

The simplest and most useful method of sludge disposal is to take advantage of it as a fertilizer for direct land application. Processed aquaculture sludge dry matter contains high levels of nitrogen and phosphorus, but negligible amounts of potassium. Nitrogen is mainly organically bound and has to be decomposed by microorganisms in order to become available for plants (Bergheim et al. 1993). When the solids content is less than 1%, solids and slurries are easily pumped and distributed. Thus, there are a variety of land application systems that can be used, including conventional irrigation via sprinklers and surface flooding. Climate constraints on crops and vegetation may require winter storage of the wastewater. Other methods include rapid infiltration of the wastewater by intermittent flooding of shallow basins in relatively coarse textured soils of high permeability. Thickened sludge (>5% solids) can be used as a soil amendment or fertilizer and applied from tanker trucks, by surface spreading with or without incorporation into the soil, and by direct injection into the soil.

COMPOSTING

(adapted from Agricultural Waste Management Field Handbook, NRCS, 1996)



Composting is the aerobic biological decomposition of organic matter. It is a natural process that is enhanced and accelerated by the mixing of aquaculture wastes with other ingredients for optimum microbial growth. Composting converts the sludge and other waste products from aquaculture into a stable organic product by converting nitrogen from the unstable ammonia to a more stable organic form. The end result is a product that is safer to use than the original sludge and when applied to land, will improve the soil fertility, tilth, and water holding capacity. In addition, composting reduces the bulk of material that needs to be spread, improves its handling properties, reduces odor, fly and other vector problems and can destroy pathogens.

Composting methods include windrow, static pile, and in-vessel. The windrow method consists of piling the compost mix into long, narrow piles or windrows. To maintain aerobic conditions, this windrow needs to be periodically turned and mixed, exposing the decomposing material to air and to keep the temperatures from getting too high. The static pile method consists of mixing the compost materials and then stacking the mixture on plastic pipe or tubing through which air can be forced or drawn. Small compost piles may not need forced ventilation, if they are highly porous or with a mix that is stacked in layers with highly porous materials. The in-vessel method involves the mixing of the materials in a reactor, building, container, or vessel. Forced ventilation may be required. This process provides a high level of control over moisture, aeration, and temperature.

Composting of aquaculture wastes requires the mixing in of amendments or bulking agents in the proper proportions to promote aerobic microbial activity and growth and achieve optimum temperatures. This mixture provides a source of energy and nutrients, moisture, and oxygen for the bacteria. The composting amendment is added to the mixture to alter the moisture content, carbon to nitrogen (C:N) ratio, or pH. Many materials are suitable for use as composting amendments, including crop residues, leaves, grass, straw, hay, sawdust, wood chips or

shredded paper, and cardboard. The bulking agent is used to improve the ability of the compost to be self-supporting and to increase porosity to allow internal air movement. Wood chips and shredded tires are two examples of bulking agents. Wood chips are an excellent bulking agent because they also alter the moisture content and C:N ratio. Figure 6.9 shows a compost bin using wood chips as both an amendment and bulking agent.

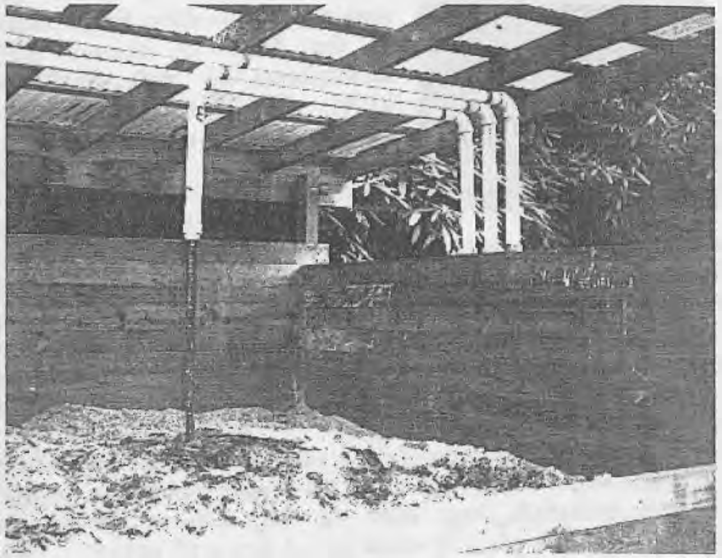


Figure 6.9 Compost bin for sludge.

Compost design requires knowledge of the characteristics of the aquaculture wastes and the amendments and bulking agents. The characteristics that are most important are moisture content, carbon content, nitrogen content and C:N ratio. The C:N ratio in the compost mixture is a critical factor for optimum microbial activity. These microbes multiply rapidly in the compost pile and consume carbon as a food source and nutrients to metabolize and build proteins. The C:N ratio of the compost mix should be maintained between 25 and 40 to 1. If the C:N ratio is low, a loss of nitrogen generally occurs through rapid decomposition and volatilization of the ammonia. If it is high, the composting time increases because the nitrogen becomes the limiting nutrient for growth. Moisture is required by the microbes to convert the carbon source to energy. Bacterial generally can tolerate moisture content as low as 12 to 15%. However, at moisture contents of less than

40%, the rate of decomposition is slow. At greater than 60% moisture, the process turns from one that is aerobic to one that is anaerobic. Anaerobic composting decomposes more slowly and produces putrid odors. The determination of the compost mix design or retype is normally an iterative process of adjusting the C:N ratio and moisture content by the addition of amendments. If the C:N ratio is out of the acceptable range, then amendments are added to adjust it.

The composting of mortalities can be an economical and environmentally acceptable method. The process produces little odor and destroys harmful pathogens. Compost bins are typically about 5 feet (1.5 m) high, 5 feet (1.5 m) deep, and 8 feet (2.4 m) cross the front. The width across the front should be sized to accommodate the equipment used to load and unload the facility. To prevent spontaneous combustion and to allow for ease of monitoring, a bin height of no more than 6 feet (1.8 m) is recommended. The depth should also be sized to accommodate the equipment used. Rapid composting of mortalities occurs when the C:N ratio is maintained between 10 and 20. This is considerably lower than what is normally recommended for more traditional composting, because much of the nitrogen in the dead animal mass is not exposed on the surface. A lower C:N ratio is necessary to ensure rapid composting with elevated temperature to destroy pathogens. The moisture content of the initial compost mixture should be between 45 and 55%, by weight, to facilitate rapid decomposition. Composting of mortalities should remain aerobic at all times throughout the process. This is easily accomplished by layering the mortalities and amendments in the mix. Layers of such high porosity material as straw, wood chips and bark allow lateral movement of air in the compost mix.

6.7 DESIGN EXAMPLE – GEOBAGS

As previously described, Omega Industries (OI) requires an engineering plan for the construction and operation of an Omega Fish Aquaculture Facility (OFAF). As a rule of thumb, 25% to 35% of the feed fed to the fish will be excreted as suspended solids (or total suspended solids, TSS) on a dry matter basis. To date, there is very little design information available for using geotextile bags for the treatment of aquaculture waste streams, as opposed to the treatment of other industrial sludge. One of the primary difficulties designing for aquaculture systems is the wide variation of the waste concentration, composition, salinity, pH, temperature, quantity and availability over the numerous systems available for water treatment designs and species grown. For example, the difference in concentration between

microscreen backwash water can vary from 500 to 2000 mg/L depending upon the screen mesh size and the frequency of backwashing. Sand filters generate and enormous quantity of low concentrated backwash water. Even with a "salt water system", the actual salinity might vary from as low as 2 ppt to 38 ppt depending upon the species being grown and their current growth phase.

Using a design spreadsheet to make an estimate of yearly feed loadings or you can make an accurate estimate of yearly feed usage by multiplying the yearly production times an average feed conversion, which yields a yearly feed rate of 52,750 kg (116,000 lbs). Using good feed management and high protein feed should yield an average of 25% solids waste or 13,185 kg (29,000lbs) of solids wastes production per year. Assuming further that the solids capture device being used (such as a micro-screen drum filter) yields a mean TSS concentration in the waste effluent of 2,000 mg/. This means that each day approximately 18.1 m^3 (4,780 gallons) of waste are generated, Table 6.9.

Table 6.9 Final Biomass, Feed Rates, and sludge volume based on 2000 mg/L sludge concentration for the Three Stage Omega Fish Production Strategy

	Feed per day	Sludge production rate per day	Volume of sludge/day m^3 (gal)
Juvenile:	20.9 kg (46 lbs)	5.2 kg (11.5 lbs)	2.6 m^3 (690 gal)
Fingerling:	44.8 kg (98.6 lbs)	11.2 kg (24.6 lbs)	5.6 m^3 (1,480 gal)
Growout:	78.8 kg (173 lbs)	19.7 kg (43.3 lbs)	9.85 m^3 (2,600 gal)
Totals	144.5	36.1	18.1

Using design criteria typically applied for other types of sludge, the following design concept is proposed. First, a moderately sized sludge sump tank is used to act as a flow equalizing tank to allow temporary storage of sludge for batch loading into the geotextile bag. This would provide a consistent flow into the geotextile bag. A simple dosing pump is used to add a predetermined concentration of the coagulation and flocculation aid. The geotextile bag would be loaded each fill cycle up to ~85% of its volume and then allowed to drain and consolidate for 8 to 12 hours. This cycle is repeated, until the final geotextile bag capacity of 85% of consolidated solids is reached, i.e., the bag is at 85% of its volume capacity at the end of one drainage cycle, typically one day. Additional drainage time between fill cycles (several days) may extend the time that a bag can be used. This of course also means additional bags are needed.

It should be noted that, geotextile bags are designed to with stand a significant bursting pressure, but studies done at Freshwater Institute (Ebeling & Riesel, 2006) showed that pressurizing the geotextile bag resulted in significant carryover of solids into the effluent stream. In addition, a rule of thumb for design is not to allow the pressure in the bags to exceed 0.33 atmospheres (5 psi) (Gaffney, 1999). Maximum pressure on multiple bags could be limited to approximately 1-2 psi by using a standpipe/overflow to direct the flow from the full bag to the next bag in line. The coagulation and flocculation aid can be mixed into the pumped flow stream with either a serpentine pipe pathway of PVC elbows or an inline mixing column. The geotextile bags would be supported on a gravel bed with a liner and drainage pipes to direct the effluent flow into a small sump. From the sump, the effluent would then be further treated for reuse in the aquaculture system or used for some other purpose, such as hydroponics.

Thus if we start with a daily production of 18.1 m^3 (4,780 gallons or 23.5 yds^3) of waste water and let the initial fill be **one fourth the design capacity** of the geotextile bag, then the bag volume should be approximately 77 m^3 (100 yds^3), or equal to a 4.5 m x 15 m (15' x 50') geotextile bag. The geobag is dosed three times a day to allow the sludge to dewater between dosings. The required batch dose load is initially collected in a large reservoir tank sized to hold 8 hours of sludge production or in this case 6 m^3 (~1600 gallons); the solids are maintained in suspension with an aeration grid. The discharges from each of the solids capture devices with the production pods are pumped to this central sump using a sump pump at each pod's solids collection device. At AST, a square PolyTank, PT-612, 24" x 24" x 18" and a Little Giant 1/3 HP Model 6-CIA submersible sump pump with a diaphragm switch is located at each of the three PBF filters (the solids collection devices at each pod) to pump the wastewater to the central sump that is used as the sludge equalizing tank.

Thus for a dosing of approximately 6 m^3 (1600 gallons) of filter backwash water, a 7.6 m^3 (2000 gallon) polyethylene, US Plastics Model 10000, 90" in diameter and 84" tall tank would work. A simple float switch can then be used to activate a submersible pump, when the level in the equalizing sump tank reaches a predetermined depth. In addition, to activating the sump pump, a dosing pump is also turned on to inject

the required coagulation/flocculation aid. A possible dosing pump is the Chem-Tech Series Model 240, a positive displacement diaphragm metering pump or the Blackstone Model BL20 Metering pump, both are self-priming and the feed rate is fully adjustable over the feed rate control operating range. Several days of coagulant/ flocculation aid is stored in either a 65 or 125 gallon US Plastics Free standing chemical tank.

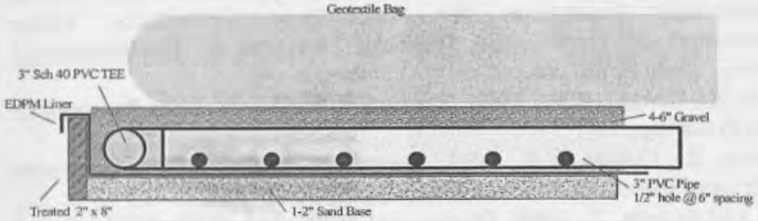


Figure 6.10 Cross-section of gravel filled, lined pad to collect effluent from geotextile bags.

To collect the supernatant from the geotextile bags, a lined gravel bed with drainage pipes is used to direct supernatant flow into a small collection sump, Fig. 6.10 above. This would be constructed out of a frame of 2x8 treated lumber 8ft by 12 ft. The EDPM liner would be protected from punctures with a thin layer of sand (1 to 2"). The drainage pipes would be constructed out of 3" Sch 40 PVC with ½" drain holes drilled every 6" along the bottom, offset from the center line by about 1". The drain lines would connect to a collector drain with 3" Sch 40 TEEs or sweep TEE's. This would then discharge into a sump tank, with a submersible sump pump with a diaphragm switch. The supernatant would be pumped into a large clean water reservoir for further treatment before being pumped into the primary water reservoir. At least three geotextile bags would be required for this design, one filling, one for overflow and one full that is aging gracefully and continuing to dewater (it may take 2 to 4 months for a bag to achieve solids contents of ~ 30 %), additional pads can be constructed adjacent to this pad and the supernatant routed to the same pump sump.

Secondary Treatment System Design

The secondary treatment system of the geotextile bag supernatant will incorporate three aspects of an engineered approach to water reclamation: bioclarification (mechanical/bio-filtration), disinfection or sterilization, and denitrification (see Fig. 6.11). All equipment is sized according to the expected volume of effluent discharge. A floating bead filter (FBF) from AST will be used for bioclarification; a UV sterilizer will be used for disinfection; and an activated carbon filter will be used

for final polishing and removal of trace organics, metals, and other potential pollutants. At some point in the near future, a commercial denitrifier could be used to reduce nitrate-nitrogen concentration. This suite of secondary treatment equipment will constitute its own recirculating loop to ensure maximum stripping of unwanted solids and nutrients prior to being reintroduced into the culture system water reservoir.

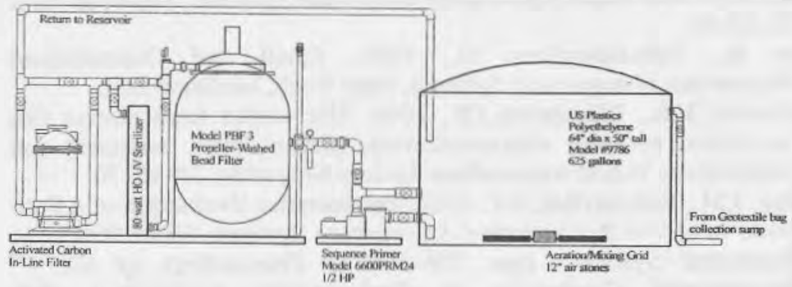


Figure 6.11 Possible configuration of the secondary treatment system.

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List of Symbols

A	cross sectional area, cm^2
D	cumulative depth of wastewater applied, m
K_s	saturated hydraulic conductivity, cm/d
L	depth of manure layer or material providing the resistance to flow, m
N	number of applications of wastewater
Q	volumetric flow rate, cm^3/d
Y	depth of wastewater applied per application event, m
β	constant used to estimate accumulation rate of manure per unit of waste water applied, m/m
ΔH	average hydraulic load imposed per application event (1/2 of treatment depth), m

Chapter I of title 40 of the Code of Federal Regulations is amended by adding part 451 to read as follows:

PART 451 — CONCENTRATED AQUATIC ANIMAL PRODUCTION POINT SOURCE CATEGORY

Subpart A—Flow-Through and Recirculating Systems Subcategory

451.10 Applicability.
451.11 Effluent limitations attainable by the application of the best practicable control technology currently available (BPT).
451.12 Effluent limitations attainable by the application of the best available technology economically achievable (BAT).
451.13 Effluent limitations attainable by the application of the best conventional technology (BCT).
451.14 New source performance standards (NSPS).

Subpart B—Net Pen Subcategory

451.20 Applicability.
451.21 Effluent limitations attainable by the application of the best practicable control technology currently available (BPT).
451.22 Effluent limitations attainable by the application of the best available technology economically achievable (BAT).
451.23 Effluent limitations attainable by the application of the best conventional technology (BCT).
451.24 New source performance standards (NSPS).

§ 451.1 General applicability.

As defined more specifically in each subpart, this Part applies to discharges from concentrated aquatic animal production facilities as defined at 40 CFR 122.24 and Appendix C of 40 CFR Part 122. This Part applies to the discharges of pollutants from facilities that produce 100,000 pounds or more of aquatic animals per year in a flow through, recirculating, net pen or submerged cage system.

§ 451.2 General definitions.

As used in this part:

(a) The general definitions and abbreviations in 40 CFR part 401 apply.

(b) *Approved dosage* means the dose of a drug that has been found to be safe and effective under the conditions of a new animal drug application.

(c) *Aquatic animal containment system* means a culture or rearing unit such as a raceway, pond, tank, net or other structure used to contain, hold or produce aquatic animals. The containment system includes structures designed to hold sediments and other materials that are part of a wastewater treatment system.

(d) *Concentrated aquatic animal production facility* is defined at 40 CFR 122.24 and Appendix C of 40 CFR Part 122.

(e) *Drug* means any substance defined as a drug in section 201(g)(1) of the
Federal Food, Drug and Cosmetic Act (21 U.S.C. 321).

(f) *Extra label drug use* means a drug approved under the Federal Food, Drug and Cosmetic Act that is not used in accordance with the approved label directions, see 21 CFR part 530.

(g) *Flow-through system* means a system designed to provide a continuous water flow to waters of the United States through chambers used to produce aquatic animals. Flow-through systems typically use rearing units that are either raceways or tank systems.

Rearing units referred to as raceways are typically long, rectangular chambers at or below grade, constructed of earth, concrete, plastic, or metal to which water is supplied by nearby rivers or springs. Rearing

units comprised of tank systems use circular or rectangular tanks and are similarly supplied with water to raise aquatic animals. The term does not include net pens.
(h) *Investigational new animal drug (INAD)* means a drug for which there is a valid exemption in effect under section 512(j) of the Federal Food, Drug, and Cosmetic Act, 21 U.S.C. 360b(j), to conduct experiments.
(i) *New animal drug application* is defined in 512(b)(1) of the Federal Food, Drug, and Cosmetic Act (21 U.S.C 360b(b)(1)).
(j) *Net pen system* means a stationary, suspended, or floating system of nets, screens, or cages in open waters of the United States. Net pen systems typically are located along a shore or pier or may be anchored and floating offshore. Net pens and submerged cages rely on tides and currents to provide a continual supply of high-quality water to the animals in production.
(k) *Permitting authority* means EPA or the State agency authorized to administer the National Pollutant Discharge Elimination System permitting program for the receiving waters into which a facility subject to this Part discharges.
(l) *Pesticide* means any substance defined as a ''pesticide'' in section 2(u) of the Federal Insecticide, Fungicide, and Rodenticide Act (7 U.S.C. 136(u)).
(m) *Real-time feed monitoring* means a system designed to track the rate of feed consumption and to detect uneaten feed passing through the nets at a net pen facility. These systems may rely on a combination of visual observation and hardware, including, but not limited to, devices such as video cameras, digital scanning sonar, or upweller systems that allow facilities to determine when to cease feeding the aquatic animals. Visual observation alone from above the pens does not constitute real-time monitoring.
(n) *Recirculating system* means a system that filters and reuses water in which the aquatic animals are produced prior to discharge. Recirculating systems typically use tanks, biological or mechanical filtration, and mechanical support equipment to maintain high quality water to produce aquatic animals.

§ 451.3 General reporting requirements.
(a) *Drugs.* Except as noted below, a permittee subject to this Part must notify the permitting authority of the use in a concentrated aquatic animal production facility subject to this Part of any investigational new animal drug (INAD) or any extralabel drug use where such a use may lead to a discharge of the drug to waters of the U.S. Reporting is not required for an INAD or extralabel drug use that has been previously approved by FDA for a different species or disease if the INAD or extralabel use is at or below the approved dosage and involves similar conditions of use.

(1) The permittee must provide a written report to the permitting authority of an INAD's impending use within 7 days of agreeing or signing up to participate in an INAD study. The written report must identify the INAD to be used, method of use, the dosage, and the disease or condition the INAD is intended to treat.

(2) For INADs and extralabel drug uses, the permittee must provide an oral report to the permitting authority as soon as possible, preferably in advance of use, but no later than 7 days after initiating use of that drug. The oral report must identify the drugs used, method of application, and the reason for using that drug.

(3) For INADs and extralabel drug uses, the permittee must provide a written report to the permitting authority within 30 days after initiating use of that drug. The written report must identify the drug used and include: the reason for treatment, date(s) and time(s) of the addition (including duration), method of application; and the amount added.

(b) Failure in, or damage to, the structure of an aquatic animal containment system resulting in an unanticipated material discharge of pollutants to waters of the U.S. In accordance with the following procedures, any permittee subject to this Part must notify the permitting authority when there is a reportable failure.

(1) The permitting authority may specify in the permit what constitutes reportable damage and/or a material discharge of pollutants, based on a consideration of production system type, sensitivity of the receiving waters and other relevant factors.

(2) The permittee must provide an oral report within 24 hours of discovery of any reportable failure or damage that results in a material discharge of pollutants, describing the cause of the failure or damage in the containment system and identifying materials that have been released to the environment as a result of this failure.

(3) The permittee must provide a written report within 7 days of discovery of the failure or damage documenting the cause, the estimated time elapsed until the failure or damage was repaired, an estimate of the material released as a result of the failure or damage, and steps being taken to prevent a recurrence.

(c) In the event a spill of drugs, pesticides, or feed occurs that results in a discharge to waters of the U.S., the permittee must provide an oral report of the spill to the permitting authority within 24 hours of its occurrence

and a written report within 7 days. The report shall include the identity and quantity of the material spilled.
(d) *Best management practices (BMP) plan.* The permittee subject to this Part must:

(1) Develop and maintain a plan on site describing how the permittee will achieve the requirements of § 451.11(a) through (e) or § 451.21(a) through (h), as applicable.
(2) Make the plan available to the permitting authority upon request.
(3) The permittee subject to this Part must certify in writing to the permitting authority that a BMP plan has been developed.

Subpart A—Flow-Through and Recirculating Systems Subcategory

§ 451.10 Applicability
This subpart applies to the discharge of pollutants from a concentrated aquatic animal production facility that produces 100,000 pounds or more per year of aquatic animals in a flow through or recirculating system.

§ 451.11 Effluent limitations attainable by the application of the best practicable control technology currently available (BPT)
Except as provided in 40 CFR 125.30 through 125.32, any existing point source subject to this subpart must meet the following requirements, expressed as practices (or any modification to these requirements as determined by the permitting authority based on its exercise of its best professional judgment) representing the application of BPT:

(a) *Solids control*
The permittee must:

(1) Employ efficient feed management and feeding strategies that limit feed input to the minimum amount reasonably necessary to achieve production goals and sustain targeted rates of aquatic animal growth in order to minimize potential discharges of uneaten feed and waste products to waters of the U.S.
(2) In order to minimize the discharge of accumulated solids from settling ponds and basins and production systems, identify and implement procedures for routine cleaning of rearing units and off-line settling basins, and procedures to minimize any discharge of accumulated solids during the inventorying, grading and harvesting aquatic animals in the production system.
(3) Remove and dispose of aquatic animal mortalities properly on a regular basis to prevent discharge to waters of the U.S., except in cases where the permitting authority authorizes such discharge in order to benefit the aquatic environment.

(b) *Materials storage*
The permittee must:

(1) Ensure proper storage of drugs, pesticides, and feed in a manner designed to prevent spills that may result in the discharge of drugs, pesticides or feed to waters of the U.S.
(2) Implement procedures for properly containing, cleaning, and disposing of any spilled material.

(c) *Structural maintenance*
The permittee must:

(1) Inspect the production system and the wastewater treatment system on a routine basis in order to identify and promptly repair any damage.
(2) Conduct regular maintenance of the production system and the wastewater treatment system in order to ensure that they are properly functioning.

(d) *Recordkeeping*
The permittee must:

(1) In order to calculate representative feed conversion ratios, maintain records for aquatic animal rearing units documenting the feed amounts and estimates of the numbers and weight of aquatic animals.
(2) Keep records documenting the frequency of cleaning, inspections, maintenance, and repairs.

(e) *Training*
The permittee must:

(1) In order to ensure the proper clean-up and disposal of spilled material adequately train all relevant facility personnel in spill prevention and how to respond in the event of a spill.
(2) Train staff on the proper operation and cleaning of production and wastewater treatment systems including training in feeding procedures and proper use of equipment.

§ 451.12 Effluent limitations attainable by the application of the best available technology economically achievable (BAT)
Except as provided in 40 CFR 125.30 through 125.32, any existing point source subject to this subpart must meet the following requirements representing the application of BAT: The limitations are the same as the corresponding limitations specified in § 451.11.

§ 451.13 Effluent limitations attainable by the application of the best conventional technology (BCT).

Except as provided in 40 CFR 125.30 through 125.32, any existing point source subject to this subpart must meet the following requirements representing the application of BCT: The limitations are the same as the corresponding limitations specified in § 451.11.

§ 451.14 New source performance standards (NSPS)

Any point source subject to this subpart that is a new source must meet the following requirements: The standards are the same as the corresponding limitations specified in 451.11.

CHAPTER 7
BIOFILTRATION

7.0 INTRODUCTION

Nitrogen is an essential nutrient for all living organisms and is found in proteins, nucleic acids, adenosine phosphates, pyridine nucleotides, and pigments. In the aquaculture environment, nitrogen is of primary concern as a component of the waste products generated by rearing fish. There are four primary sources of nitrogenous waste: ammonia, urea, uric acid, and amino acids excreted by the fish, organic debris from dead and dying organisms, uneaten feed and feces, and nitrogen gas from the atmosphere. In particular, fish expel various nitrogenous waste products through gill diffusion, gill cation exchange, urine, and feces. The decomposition of these nitrogenous compounds is particularly important in intensive recirculating aquaculture systems (RAS) because of the toxicity of ammonia, nitrite, and to some extent, nitrate. The process of ammonia removal by a biological filter is called nitrification and consists of the successive oxidation of ammonia to nitrite and finally to nitrate. The reverse process is called denitrification and is an anaerobic process where nitrate is converted to nitrogen gas with an intermediary step of nitrite. Although not normally employed in commercial aquaculture facilities today, the denitrification process (Chapter 9 Denitrification) is becoming increasingly important, especially in marine systems, as stocking densities increase and water exchange rates are reduced, resulting in excessive levels of nitrate in the culture system. Recently, zero-exchange management systems have been developed based on heterotrophic bacteria and promoted for the intensive production of marine shrimp and tilapia. In these systems, heterotrophic bacterial growth is stimulated through the addition of organic carbonaceous substrate. At high organic carbon to nitrogen (C/N) feed ratios, heterotrophic bacteria assimilate ammonia-nitrogen directly from the water replacing the need for an external fixed film biofilter.

7.1 NITRIFICATION (AUTOTROPHIC BACTERIA)

Ammonia is produced as the major end-product of protein catabolism and is excreted by fish primarily as unionized ammonia across their gills. Ammonia, nitrite, and nitrate are all highly soluble in

water. Ammonia exists in two forms: un-ionized NH_3, and ionized NH_4^+. The relative concentration of each of these forms of ammonia in the water column is primarily a function of pH, temperature, and salinity (Anthonisen et al. 1976). The sum of the two ($NH_4^+ + NH_3$) is called total ammonia or simply ammonia. It is common in chemistry to express inorganic nitrogen compounds in terms of the nitrogen they contain, i.e., NH_4^+–N (ionized ammonia nitrogen), NH_3–N (un-ionized ammonia nitrogen), NO_2–N (nitrite nitrogen), and NO_3–N (nitrate nitrogen) (Table 7.1). This allows for easier computation of total ammonia-nitrogen (TAN = NH_4^+–N + NH_3–N) and easy conversion between the various stages of nitrification.

Table 7.1 Concentrations of Nitrogenous Compounds when Normalized to the Molecular Weight of Nitrogen

Nitrogen based name	Nitrogen based concentration of 1 mg/L	Equivalent concentration of compound in mg/L
Ammonia-nitrogen	NH_3-N	1.21 NH_3
Ammonium-nitrogen	NH_4^+-N	1.29 NH_4^+
Total Ammonia-nitrogen (TAN)	TAN	1.21 NH_3 or 1.29 NH_4^+
Nitrite-nitrogen	NO_2^--N	3.29 NO_2^-
Nitrate-nitrogen	NO_3^--N	4.43 NO_3^-

An increase in pH or temperature increases the proportion of the un-ionized form of ammonia nitrogen, Table 2.5. For example, at 20°C and a pH of 7.0, the mole fraction of un-ionized ammonia is only 0.4%, but increases to 80% at a pH of 10. Un-ionized ammonia is toxic to fish at low concentrations, with 96-hour LC_{50}'s (LC is lethal concentration) varying widely by species from as low as 0.08 mg/L NH_3-N for pink salmon to 2.2 mg/L NH_3-N for common carp (Lawson, 1995). For long-term exposure, the concentrations of un-ionized ammonia is dependent upon the species and the culture temperature, but as a general rule of thumb should be kept below 0.1 to 0.05 mg/L NH_3–N.

Nitrite is an intermediate product in the process of nitrification of ammonia to nitrate. Although it is usually converted to nitrate as quickly as it is produced, lack of biological oxidation of the nitrite will result in elevated nitrite levels that can be toxic to the fish. High levels of nitrite are also indicative of biofilter impending failure and should always be addressed. The toxicity of nitrite is due to its effect on the oxygen carrying capacity of the blood hemoglobin. When it enters the bloodstream, nitrite oxidizes the iron in the hemoglobin molecule from

the ferrous state to the ferric state. The resulting product is called methemoglobin, which has a characteristic brown color, yielding the common name brown-blood disease (Tomasso et al. 1979, Colt and Tchobanoglous, 1976). The amount of nitrite entering the blood depends on the ratio of nitrite to chloride in the water. Chloride levels can be increased to lessen the effects of nitrite toxicity. At least a 20:1 ratio of chloride to nitrite-nitrogen (Cl: NO_2-N) is recommended for channel catfish in ponds, tilapia, and rainbow trout (Tucker and Robinson, 1990). Chloride levels can be increased by adding ordinary salt (sodium chloride) or calcium chloride.

Nitrate is the end-product of nitrification and is the least toxic of the nitrogen compounds, with 96-h LC_{50} values in freshwater usually exceeding 1000 mg NO_3-N /L (Colt and Tchobanoglous, 1976). In recirculating systems, nitrate levels are usually controlled by daily water exchanges. In systems with low water exchange or high hydraulic retention times, denitrification has become increasingly important as a control measure. With the increase in marine systems, the need for denitrification is important due to the high hydraulic retention times in these systems and the high cost of formulating salt water and disposing of it.

Biological filtration can be an effective means of controlling ammonia; as opposed to water flushing to control ammonia levels. There are two phylogenetically distinct groups of bacteria that collectively perform nitrification. These are generally categorized as chemosynthetic autotrophic bacteria because they derive their energy from inorganic compounds as opposed to heterotrophic bacteria that derive energy from organic compounds (Hagopian and Riley, 1998). Ammonia oxidizing bacteria (AOB) obtain their energy by catabolizing un-ionized ammonia to nitrite and include bacteria of the genera *Nitrosomonas, Nitrosococcus, Nitrosospira, Nitrosolobus*, and *Nitrosovibrio*. Nitrite oxidizing bacteria (NOB) oxidize nitrite to nitrate, and include bacteria of the genera *Nitrobacter, Nitrococcus, Nitrospira*, and *Nitrospina*. Nitrifying bacteria are primarily obligate autotrophs, which consume carbon dioxide as their primary inorganic carbon source, and obligate aerobes, which require oxygen to grow (Hagopian and Riley, 1998). In biofilters, the nitrifying bacteria usually coexist with heterotrophic microorganisms such as heterotrophic bacteria, protozoa, and micrometazoa, which metabolize degradable organic compounds. Heterotrophic bacteria grow significantly faster than nitrifying bacteria and will prevail over nitrifying bacteria in competition for space and oxygen in biofilters, when concentrations of dissolved and particulate organic matter are high. For this reason, it is imperative that the source

water for biofilters be as clean as possible with minimal concentration of total organic solids.

Nitrification is a two-step process, where ammonium is first oxidized to nitrite and then nitrite is oxidized to nitrate. The two steps in the reaction are normally carried out sequentially. Since the first step has a higher kinetic reaction rate than the second step, the overall kinetics are usually controlled by ammonia oxidation and as a result, there should be no appreciable amount of nitrite accumulation. Equations 7.1 thru 7.6 show the basic chemical conversions occurring during oxidation by *Nitrosomonas* and *Nitrobacter* and the overall oxidation reaction (U.S. EPA, 1993; Ebeling et al, 2006).

Typical startup characteristics for bringing a new biological filter system up to full capacity are shown in Fig. 7.1. Note that ammonia concentration peaks at 14 days followed by a nitrite peak at 28 days and nitrate accumulation begins after 21 days. Pre-seeding a biological filter with both ammonia and nitrite can accelerate this process. For safety, in a new system, you should observe a drop in nitrite before stocking fish as an indication that the biological filtration process is fully activated.

Nitrosomonas

$$NH_4^+ + 1.5\,O_2 \rightarrow NO_2^- + 2\,H^+ + H_2O \qquad (7.1^a)$$

Nitrobacter

$$NO_2^- + 0.5\,O_2 \rightarrow NO_3^- \qquad (7.2)$$

Overall

$$NH_4^+ + 2\,O_2 \rightarrow NO_3^- + 2\,H^+ + H_2O \qquad (7.3)$$

The overall reaction of nitrification and cell biomass formation can also be written as (Haug and McCarty, 1972):

Nitrosomonas

$$55\,NH_4^+ + 5\,CO_2 + 76\,O_2 \rightarrow$$
$$C_5H_7NO_2 + 54\,NO_2^- + 52\,H_2O + 109\,H^+ \qquad (7.4)$$

Nitrobacter

$$400\,NO_2^- + 5\,CO_2 + NH_4^+ + 195\,O_2 + 2\,H_2O \rightarrow$$
$$C_5H_7NO_2 + 400\,NO_3^- + H^+ \qquad (7.5)$$

[a] Symbols are defined at the end of the Chapter.

Overall (Ebeling, et al., 2006a)

$$NH_4^+ + 1.83\,O_2 + 1.97\,HCO_3^- \rightarrow$$
$$0.0244\,C_5H_7O_2N + 0.976\,NO_3^- + 2.90\,H_2O + 1.86\,CO_2 \qquad (7.6)$$

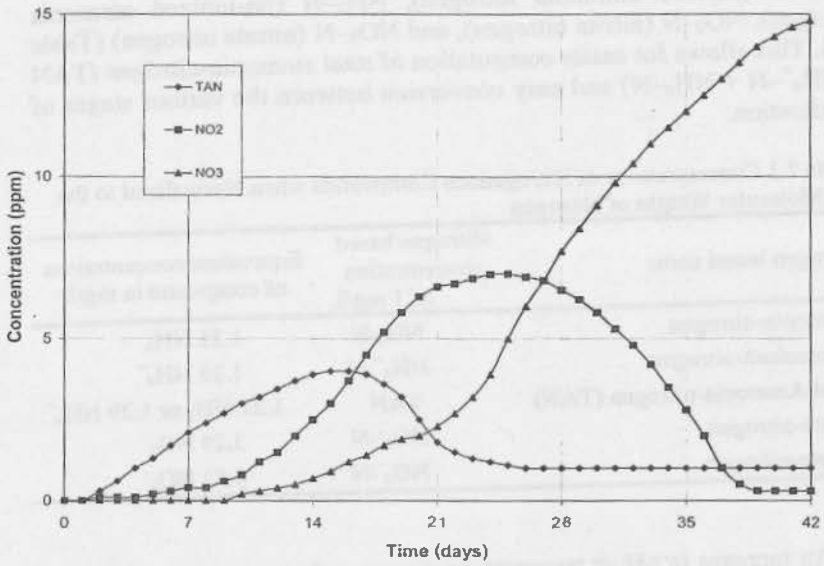

Figure 7.1 Typical startup curve for a biological filter.

Using this stoichiometric relationship (Eq. 7.6), for every gram of ammonia-nitrogen converted to nitrate-nitrogen, 4.18 g of dissolved oxygen, and 7.05 g of alkalinity (1.69 g inorganic carbon) is consumed and 0.20 g of microbial biomass (0.105 g organic carbon) and 5.85 gm of CO_2, (1.59 g inorganic carbon) is produced. It should be noted that both the consumption of oxygen and alkalinity is less than that which normally reported, 4.57 g of O_2 and 7.14 g of alkalinity for every gram of ammonia-nitrogen converted, because in this equation some of the ammonia-nitrogen is converted to biomass. Traditionally, this biomass has not been included in the stoichiometric relationship because it is minor in comparison to the other factors. Alkalinity should be maintained at 100 to 150 mg/L $CaCO_3$ through the addition of chemicals containing hydroxide, carbonate, or bicarbonate ions. Sodium bicarbonate (baking soda) is usually used since it is relatively safe, easy to obtain and dissolves rapidly and completely in water. As a rule of

thumb, for every kg of feed fed, approximately 0.25 kg of sodium bicarbonate (4.7 g alkalinity as $CaCO_3$ per g TAN assuming 35% P feed) is needed to replace the lost alkalinity consumed during nitrification (Loyless and Malone, 1997). Table 7.2 summarizes the stoichiometry for metabolism of 1 gram of ammonia-nitrogen by autotrophic bacterial, including the consumption and production of organic and inorganic carbon.

Table 7.2 Stoichiometry for Autotrophic Bacteria Metabolism of 1.0 g NH_4^+-N (Ebeling, et al., 2006)

Consumables	Stoichiometry	Consumes (g)	$C_{organic}$ (g)	$C_{inorganic}$ (g)	N (g)
NH_4^+-N		1.0	-----	-----	1.0
Alkalinity	7.05 g Alk/ g N	7.05	-----	1.69	-----
Oxygen	4.18 g O_2/ g N	4.18	-----	-----	-----
Products	**Stoichiometry**	**Yields (g)**	**$C_{organic}$ (g)**	**$C_{inorganic}$ (g)**	**N (g)**
VSS_A	0.20 g VSS_A / g N	0.20	0.106	-----	0.024
NO_3^--N	0.976 g NO_3^--N /g N	0.976	-----	-----	0.976
CO_2	5.85 g CO_2/ g N	5.85	-----	1.59	-----

In the autotrophic nitrification process, as opposed to heterotrophic processes, very small amounts of bacterial biomass are produced. Moreover, because of the relatively slow maximum growth rate for the nitrifiers in a suspended-growth process, it becomes very easy to 'wash-out' the nitrifying bacteria as opposed to a fixed-film system. This is particularly true if there is no sludge recycling that returns the bacteria back into the culture system. In addition, there is a significant amount of alkalinity consumed (7.05 g (as $CaCO_3$)/g N) and high levels of carbon dioxide produced (5.85 g CO_2 /g N). For water with low initial alkalinity this can be a significant problem, requiring the addition of alkalinity, in the form of sodium bicarbonate, lime, sodium hydroxide, to maintain an adequate alkalinity (100 to 150 mg/L as $CaCO_3$), especially for systems with limited water exchange. If alkalinity consumption is not compensated for by supplementation, the system pH will drop. Lowering pH will result in an inorganic carbon species shift from bicarbonate to dissolved carbon dioxide, and this increase in dissolved carbon dioxide could affect some aquaculture species. Although CO_2 concentration can be controlled with gas stripping towers, significant energy is required for pumping both the water and air through these systems. The end-product of the nitrification reaction is nitrate-nitrogen, which is not normally toxic at moderate levels in aquaculture production systems, e.g., several hundred mg/L.

The ratio of the biodegradable organic carbon to the nitrogen available for nitrification is argued to be one of the critical factors affecting the design and operation of a nitrification system (U.S. EPA, 1993). Heterotrophic bacteria have a maximum growth rate significantly higher than nitrifiers, 5 day^{-1} compared to 1 day^{-1} (U.S. EPA, 1993), thus in systems with even relatively modest C/N ratios, the heterotrophs are capable of outperforming and significantly inhibiting nitrification. Zhu and Chen (2001) demonstrated the effect of sucrose on the nitrification rate of biofilters under steady-state conditions. They determined that at carbon/nitrogen ratios from 1.0 to 2.0 (mass of organic C to mass of N), there was a 70% reduction of total ammonia-nitrogen removal rate as compared to C/N = 0. The data suggested that the nitrification rate decreased with an increase in the organic concentration, but the impact became less pronounced when the carbon concentration became sufficiently high.

Biological treatment processes employ bacteria that grow either attached to a surface (fixed films) or that grow suspended in the water column (Wheaton et al. 1994). Almost all recirculating systems use fixed-film bioreactors, where the nitrifying bacteria grow on either a wetted or submerged media surface. The ammonia removal capacity of biological filters is largely dependent upon the total surface area available for growth of the nitrifying bacteria. For maximum efficiency, the media used must balance a high specific surface area, i.e., surface per unit volume, with appreciable voids ratio (pore space) for adequate hydraulic performance of the system. The media used in the biofilters must be inert, non-compressible, and not biologically degradable. Typical media used in aquaculture biofilters are sand, crushed rock or river gravel, or some form of plastic or ceramic material shaped as small beads, or large spheres, rings, or saddles. Biofilters must be carefully designed to avoid oxygen limitation or excessive loading of solids, biochemical oxygen demand, or ammonia.

7.2 NITRIFICATION (MICROBIAL FLOC)

Two major categories of microbial floc systems are found in aquaculture. The first is photoautotrophic systems, often referred to as 'greenwater systems', which use natural blooms of algae to control nitrogen. Recently, a second microbial floc system has found commercial success where heterotrophic bacterial growth is stimulated through the addition of organic carbonaceous substrate. At high carbon to nitrogen (C/N) feed ratios, heterotrophic bacteria will assimilate ammonia-nitrogen directly into cellular protein

Photoautotrophic (algal based systems)

Conventional aquaculture ponds rely on the use of algal biosynthesis for the removal of the majority of inorganic nitrogen. The major disadvantage of algal based systems are the wide diurnal variations in dissolved oxygen, pH and ammonia-nitrogen and the long term changes in algal density and frequent 'die-offs' (Burford, et al. 2003). Unmanaged algal populations in conventional ponds typically can fix 2-3 g carbon/m^2-day. High rate mixed ponds that are well managed can yield higher rates, 10 -12 g carbon/m^2 day (Brune, et al., 2003).

The biosynthesis of saltwater algae can be described in general by the following stoichiometric relationships (Stumm and Morgan, 1996) for ammonia as the nitrogen source:

$$16NH_4^+ + 92\ CO_2 + 92\ H_2O + 14\ HCO_3^- + HPO_4^{2-} \rightarrow C_{106}H_{263}O_{110}N_{16}P + 106\ O_2 \quad (7.7)$$

Or, for nitrate as the nitrogen source:

$$16\ NO_3^- + 124\ CO_2 + 140\ H_2O + HPO_4^{2-} \rightarrow C_{106}H_{263}O_{110}N_{16}P + 138\ O_2 + 18\ HCO_3^- \quad (7.8)$$

where $C_{106}H_{263}O_{110}N_{16}P$ represents the stoichiometric formula for seawater algae.

Note that 3.13 g of alkalinity (as $CaCO_3$) is consumed for every gram of ammonia-nitrogen consumed in the first relationship and 4.02 g of alkalinity (as $CaCO_3$) is produced for every gram of nitrate-nitrogen consumed in the second. Using these stoichiometric relationships, for every gram of ammonia-nitrogen converted to algal biomass, 18.07 g of carbon dioxide is consumed and for every gram of nitrate-nitrogen used 24.4 g of carbon dioxide. Correspondently, 15.14 g and 19.71 g of O_2 are

produced respectively per gram of ammonia-nitrogen and per gram of nitrate-nitrogen. Finally, a significant quantity of algal biomass, 15.85 g is generated per gram of either ammonia or nitrate nitrogen. Table 7.3 summarizes the stoichiometry, including the consumption and production of inorganic and organic carbon.

Table 7.3. Stoichiometry Photoautotrophic Algal Metabolism of 1.0 g NH_4^+-N (Ebeling et al., 2006)

Consumables	Stoichiometry	Consumes (g)	$C_{organic}$ (g)	$C_{inorganic}$ (g)	N (g)
NH_4^+-N		1.0	-----	-----	1.0
Carbon Dioxide	18.07 g CO_2/ g N	18.07	-----	4.93	-----
Alkalinity	3.13 g Alk/ g N	3.13	-----	0.75	-----
Products	**Stoichiometry**	**Yields (g)**	**$C_{organic}$ (g)**	**$C_{inorganic}$ (g)**	**N (g)**
VSS_{Algae}	15.85 g VSS_A / g N	15.85	5.67	-----	1.0
O_2	15.14 g O_2/ g N	15.14	-----	-----	-----

Microbial Floc (Heterotrophic Bacteria)

The removal of ammonia by heterotrophic bacteria can be described in general by the following stoichiometric relationships (Ebeling, et al., 2006a) for ammonia as the nitrogen source:

$$NH_4^+ + 1.18\ C_6H_{12}O_6 + HCO_3^- + 2.06\ O_2 \rightarrow C_5H_7O_2N + 6.06\ H_2O + 3.07\ CO_2 \quad (7.9)$$

This equation predicts that for every gram of ammonia-nitrogen converted to microbial biomass, 4.71 g of dissolved oxygen and 3.57 g of alkalinity (0.86 g inorganic carbon) and 15.17 g carbohydrates (6.07 g organic carbon) are consumed. Also 8.07 g of microbial biomass (4.29 g organic carbon) and 9.65 g of CO_2 (2.63 g inorganic carbon) are produced. Note the oxygen demand is slightly higher, the alkalinity requirement about half and the CO_2 production almost 75% greater than the corresponding reaction for nitrification. Most importantly, the microbial biomass production is 40 times greater than the biomass generated from the nitrification process: 8.07 g versus 0.20 g. Table 7.4

summarizes the stoichiometry for the heterotrophic pathways for ammonia-nitrogen conversion.

Table 7.4. Stoichiometry of Heterotrophic Bacteria Metabolism of 1.0 g NH_4^+-N with Carbohydrate as Supplemental Carbon (Ebeling et al., 2006)

Consumables	Stoichiometry	Consumes (g)	$C_{organic}$ (g)	$C_{inorganic}$ (g)	N (g)
NH_4^+-N		1.0	-----	-----	1.0
$C_6H_{12}O_6$	15.17 g Carbs/ g N	15.17	6.07	-----	-----
Alkalinity	3.57 g Alk/ g N	3.57	-----	0.86	-----
Oxygen	4.71 g O_2/ g N	4.71	-----	-----	-----
Products	**Stoichiometry**	**Yield (g)**	**$C_{organic}$ (g)**	**$C_{inorganic}$ (g)**	**N (g)**
VSS_H	8.07 g VSS_H / g N	8.07	4.29	-----	1.0
CO_2	9.65 g CO_2/ g N	9.65	-----	2.63	-----

Several aspects are important in the overall heterotrophic bacterial reaction. Paramount is the extremely large amount of bacterial biomass produced by this reaction, compared to the autotrophic reaction. Thus, some form of solids management to remove excess TSS is required. A second issue is the modest amount of alkalinity consumed (3.57 g/g TAN) and the resulting high levels of carbon dioxide produced (9.65 g/g TAN). For water with low initial alkalinity, this will generally still require the addition of carbonate, usually in the form of sodium bicarbonate to maintain reasonable alkalinity (100 to 150 mg/L as $CaCO_3$), especially for systems with limited water exchange. As a result, zero-exchange production systems that rely on suspended or attached heterotrophic bacteria usually show a modest decrease in alkalinity, large suspended solids production, and high CO_2 levels. Finally, there should be no production of nitrite-nitrogen, or nitrate-nitrogen in a pure heterotrophic system.

> **"Rule of Thumb"**
> For each 1 gram of ammonium-N nitrified in a heterotrophic driven process
> 4.71 g of Oxygen
> &
> 7.05 g as $CaCO_3$ or 0.141 equiv. of Alkalinity are required.
> *(50 g as $CaCO_3$= 1.00 eq. of Alkalinity)*

7.3 Impact of Water Quality Factors on Nitrification

Nitrification Kinetics

The rate of ammonia or nitrite oxidation is strongly dependent on the concentration of these nutrients in the bulk solution. In a pure culture, the nitrification rate can be expressed as a Monod-type expression (Srna and Baggaley, 1975; Rittman and McCarty, 1980; Zhu and Chen, 1999, 2002,; Chen et al., 2006):

$$R = \frac{R_{max}\,S}{(K_s + S)} \tag{7.10}$$

where R: substrate removal rate (g/m^2 day); R_{max}: maximum substrate removal rate (g/m^2 day); S: limiting substrate concentration (mg/L, usually ammonia or nitrite, sometimes dissolved oxygen; K_s: half saturation constant (mg/L).

This equation can be used to describe the rate of ammonia or nitrite removal, assuming no other substrate is limiting, such as dissolved oxygen. Two important characteristics of this equation are that at sufficiently high substrate concentration (ammonia > 2 mg/L), the substrate removal becomes a zero-order expression, i.e. a constant value. And second, at sufficiently low substrate concentrations (ammonia < 1 mg/L), the relationship becomes linear or a 1st order equation, i.e. directly proportional to the substrate concentration.

The nitrification rate in the biofilter is a constant balance between the demand by the AOB and NOB for nutrients for their growth and well-being and the supply of these nutrients determined by their bulk concentration and diffusion rate into the biofilm. Chen, et al., (2006) classified the more than 20 physical, chemical, and biological factors that influence the rate of nitrification into three major categories. The first includes those factors that directly influence the microbes that make up

the biofilm, such as pH, temperature, alkalinity, and salinity. The second include those that affect the supply of nutrients for the microbes such as substrate concentration (ammonia), dissolved oxygen (DO) and mass transport of nutrients controlled by the mixing/turbulence in the bioreactor. Finally, the third category includes those that have impact on both the growth and nutrient supply, such as competition for nutrients and space with heterotrophic bacteria.

pH

The effect of pH on the nitrification rate for biofilters has been researched for more than sixty years, yet there is a wide range in reported pH optima (Biesterfeld et al., 2001). This suggests that the history and condition under which the bacteria are cultured may affect their response to pH (Kaiser & Wheaton, 1983). The literature suggests that the optimum range of pH for nitrification can range from 7.0 to 9.0 (Haug and McCarty, 1972; Chen, et al., 2006). The optimum pH for *Nitrosomonas* ranges from 7.2 to 7.8 (Loveless & Painter, 1968, Antoniou et al. 1990) and from 7.2 to 8.2 for *Nitrobacter*. Nitrifying biofilters have been operated over a much broader range from 6 to 9, due to the adaptation of the bacteria in a filter to actual operating conditions. It is probably a good idea to maintain pH near the lower end of the optimum pH for the nitrifying bacteria to minimize ammonia stress on the cultivated fish species. In addition, rapid changes in pH of more than 0.5 to 1.0 units over a short time span will stress the filter and require time for adaptation to the new environmental conditions.

Temperature

Temperature plays a significant role in the nitrification reaction rate in suspended growth systems as it does in all chemical and biological kinetic reactions, although limited research is available to quantify the effects of temperature on fixed film nitrification rates (Okey and Albertson, 1989). Zhu and Chen (2002) studied the impact of temperature on nitrification rates in laboratory experiments, mathematical modeling, and sensitivity analysis. Their studies showed that the impact of temperature on the nitrification rate for fixed film nitrification was less than predicted by the van't Hoff-Arrhenius equation. More specifically, Zhu and Chen showed that in the case of no oxygen limitation, temperatures from 14 to 27 °C had no significant impact on the nitrification rate of a fixed film bioreactor. Malone and Pfeiffer (2006) reported that although originally assumed to be an

important factor in biofilter design, temperature is increasingly being viewed as a minor factor in controlling biofilter carrying capacities.

Other researchers have determined a small but significant impact of temperature on nitrification rates. In particular, Wortman and Wheaton (1991) developed the Eq. 7.11 to predict relative nitrification rates, R, over the range of their data (7 to 35°C).

$$R = 140 + 8.5\ T(°C) \tag{7.11}$$

For example, the nitrification rates at 17°C would only be 77% of the rates obtained at 27°C, or a 23% reduction in rate. This is less severe than the 50% reduction that would be predicted based upon Q-10 (Arrhenius relationship) effect for a 10°C drop in water temperature.

There is a wide range of optimum temperatures reported for nitrification (Jones and Morita, 1985; Chen et al., 2006), suggesting that nitrifying bacteria are able to adapt to a wide range of environmental temperature, if acclimated slowly. In practical application, however, the temperature at which the biofilter operates is normally determined by the requirements of the species being cultured, not by the needs of the biofilter bacteria.

Alkalinity

Alkalinity is a measure of the buffering capacity of an aquatic system. From the relationships above, it was determined that for every gram of ammonium-nitrogen reduced to nitrate-nitrogen, 7.05 grams of alkalinity is consumed. This loss of alkalinity is easily made up by the addition of sodium bicarbonate, referred to commonly as baking soda ($NaHCO_3$), or other bicarbonate supplement. A rule-of-thumb is 0.25 lbs of baking soda per pound of feed, 0.25 kg per 1 kg (Loyless and Malone, 1997). Nitrification is an acid-forming process, and if the biofilter system's water is poorly buffered, the system pH will decline and affect the biofilter performance.

Figure 7.2 shows dramatically the impact of low alkalinity on nitrification. This was a short, simple study done one summer by one of the authors when the 'living was easy' with a well acclimated biofilter. Ammonia in the form of ammonia chloride was added daily and in addition baking soda until Day 55 to maintain a constant alkalinity. The impact of not adding baking soda was a steady decline in alkalinity and a significant increase in ammonia-nitrogen (Day 62) when the alkalinity fell below 100 mg/L as $CaCO_3$. When the alkalinity was increased above

150 mg/L, nitrification resumed and ammonia-nitrogen quickly dropped to very low levels.

Note that the alkalinity consumption value of 7.05 g alkalinity (or 7.14 g in absence of heterotrophic growth) per g of ammonium-N nitrified is not accounting for the alkalinity being added when fish secrete ammonia (NH_3) across their gills (paper on this subject under construction by authors) into the same water that has the nitrification system. Ammonia (NH_3) acts as a base when added to the water (can take on an H^+ ion). So, starting from the fish excreting NH_3 which then is protonated with a hydrogen ion in the water column (absorbing acid), the net alkalinity consumption is only half that when you start with ammonium (NH_4^+), or 3.57 g of alkalinity (one equivalent) per g of ammonia-N nitrified. This is described in more detail in Chapter 9 Denitrification.

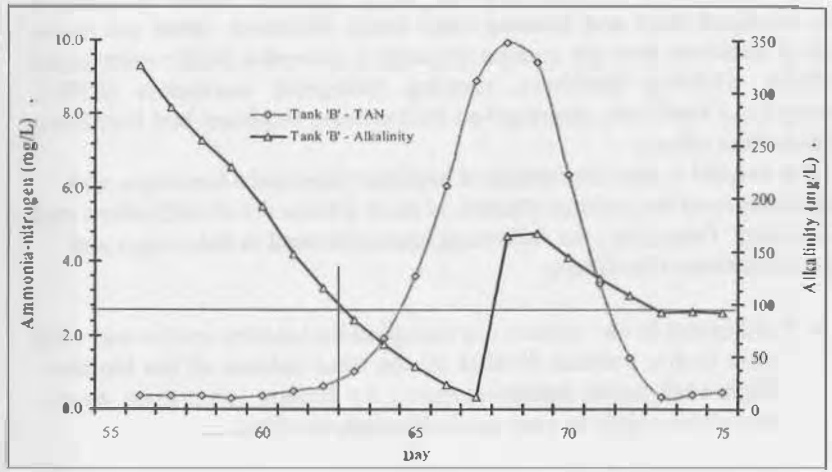

Figure 7.2 Impact of alkalinity on nitrification.

SALINITY

There is very limited information on the impact of salinity on nitrification. Salinity is similar to both temperature and pH, in that nitrifying bacteria can acclimate to almost any salinity range, given sufficient time. Chen, et al. (2006) reported that many engineering companies and pilot scale long term experiments with fresh and marine water recirculation systems suggest that the average removal rate is reduced by approximately 37% in salt water compared to fresh water. Rusten, et al. (2006) reported that data from commercial fish farms operating at a salinity of 21-24 ppt, indicated that the nitrification rate was approximately 60% of what would be expected in a freshwater system for Moving Bed BioReactors. Numerous researchers, including the authors, have observed that it takes significantly longer to fully acclimate a biofilter in salt water than in fresh water. Abrupt changes in salinity of greater than 5 g/L, will shock nitrifying bacteria and decrease the reaction rate for both ammonia-nitrogen and nitrite-nitrogen removal (Hochheimer, 1990).

AMMONIA

The ammonia concentration itself will directly affect the nitrification rate. In general, as ammonia concentration increases the biofilter performance increases proportionately over some limited range of concentrations. This linear, proportional relationship exists from very low concentrations to between 2 and 3 mg/L TAN. This proportional increase will at some point decrease, and then eventually level off to a constant removal rate. This phenomenon is shown graphically in Fig. 7.3 (Ebeling and Wheaton, 2006). There is some evidence in the literature that at extremely high concentrations of ammonia and nitrite and much above any expected concentrations that will be seen in aquaculture applications, accumulating ammonia will become inhibiting to nitrification (Anthonisen et al., 1976; Carrera et al., 2004).

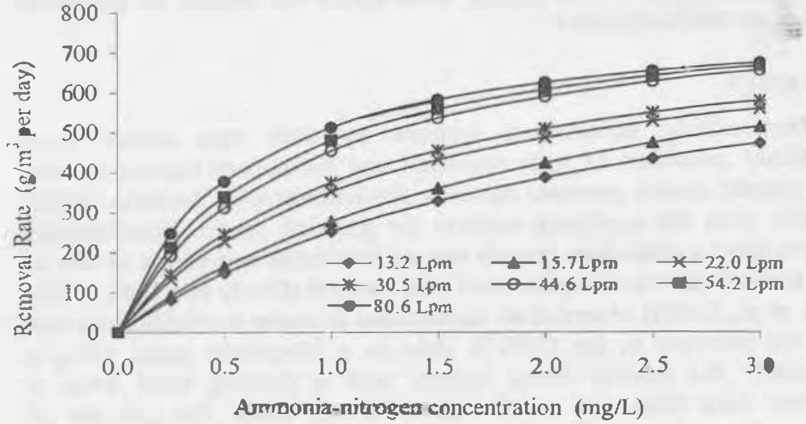

Figure 7.3 Nitrification rate as affected by water column concentration and flow rate for a bubble-washed bead filter (Ebeling and Wheaton, 2006).

DISSOLVED OXYGEN

Oxygen can become the rate-limiting factor in certain biofilters, because of the low levels in the influent and the competing demands of the heterotrophic bacteria. For every gram of ammonia-nitrogen oxidized to nitrate-nitrogen, 4.57 g of oxygen is required. Knowles et al. (1965) studied nitrification with a mixed culture reactor and reported that DO affected the growth rate of *Nitrosomonas* very little at DO levels above 2.0 mg/L, but *Nitrobacter* exhibited a reduced growth rate at DO levels of less than 4 mg/L. Wheaton (1985) and Malone et al. (1998) state that biofilter effluent levels of at least 2 mg/L of oxygen are probably adequate to maintain maximum nitrification rates.

TURBULENCE

Turbulence affects the thickness of the stagnant water film covering the bacteria and thus the transfer rate of the nutrients from the bulk liquid into the biofilm. This affect is demonstrated in Fig. 7.3, where the ammonia removal rate is seen to increase as the flow rate through the bubble-wash bead filter increases (Ebeling and Wheaton, 2006). Currently there is limited information on the impact of turbulence and the design role it plays in improving the nitrification rate in biofilters. Excessive shear (high water velocity) or abrasion (sand particles) would be assumed to have a negative impact on biofilm growth and film thickness. Zhu and Chen (2001) investigated the impact of Reynolds number on biofilm kinetics.

ORGANICS

Recirculating aquaculture systems by their very nature have significant quantities of both dissolved and particulate organic matter. This organic matter provides substrate for heterotrophic bacteria, which compete with the nitrifying bacteria for growing space. Heterotrophic bacteria have a maximum growth rate of five times and yields of two to three times that of autotrophic nitrifying bacteria (Grady and Lim, 1980). Chen, et al., (2006) observed an exponential decrease in nitrification rate with the increase in the COD/N ratio in a laboratory study using a chemically fed reactor series system with a floating bead filter, a fluidized sand filter, and a submerged biocube filter. The message of these studies is that organics, i.e. solids, need to be removed immediately from the recirculating aquaculture system.

7.4 BIOFILTERS

There is considerable debate (and significant competition) as to the most appropriate biological filter technology for intensive aquaculture applications. The task is further complicated by the wide variety of water quality requirements and environmental conditions displayed by recirculating aquaculture systems. An ideal biofilter would maximize media specific surface area and remove 100% of the inlet ammonia concentration, generate very little nitrite, maximize oxygen transfer, require a relatively small footprint, use inexpensive media, have minimal head loss, require very little maintenance to operate, and would not capture solids. Unfortunately, there is no one biofilter type that meets all of these ideals, each biofilter has its own strength and weaknesses and areas of best application. Currently large scale commercial recirculating systems have been moving towards using granular filters (expanded beds, fluidized beds and floating bead beds). However, there are many types of biofilters that are commonly used in intensive RAS: submerged biofilters, trickling biofilters, rotating biological contactors (RBC), floating bead biofilters, moving bed bioreactors, fluidized-bed biofilters, and countless others.

It is helpful in any discussion of biofilter principal advantages and disadvantages of the various choices to have a basic set of definitions and terminology. Generally, the following terms are used in the design and characterization of biofilters:

- **Void** space is the volume not occupied by biofilter media and void ratio is that volume divided by the total volume of the biofilter. High void ratios reduce clogging by having large open spaces that allow solids to pass easily through the filter.

- **Cross-sectional area** refers to the area of the filter bed looking in the direction of the water flow. Filter top area is usually one of the last parameters selected in the filter design, to yield a desired hydraulic loading rate.

- **Hydraulic loading rate** is the volume of water pumped through the biofilter per unit of cross-sectional area of the filter per unit of time, typically expressed as gpm/ft^2 or m^3/m^2 day. There is usually both a minimum and a maximum hydraulic loading rate for biofilters.

- **Specific surface area** is the surface area of the media per unit volume. The higher the specific surface area of a media, the more bacteria can grow on a unit volume and the greater the total ammonia removal per unit volume of filter. The media size, void ratio, and specific surface area are all interrelated, the smaller the size, the larger the specific surface area and the smaller the void ratio.

Table 7.5 Total Ammonia-N (TAN) Assimilation Rates for Biofilters Based on Volumetric and Areal TAN Conversion Rates and Hydraulic Loading Rate

Media Type	TAN Conversion Based On	TAN Conversion Rate (15 to 20 °C)	TAN Conversion Rate (25 to 30 °C)	Hydraulic Loading $m^3/m^2{*}d$ (gpm/ft^2)
Trickling or RBC (100–300 m^2/m^3)	Surface area of media	0.2 to 1.0 g/m^2 per day	1.0 to 2.0 g/m^2 per day	100–800 (1.7–13.8) (Kamstra et al. 1998)
Granular (bead/ sand) (>500 m^2/m^3)	Volume of media	0.6 to 0.7 kg/m^3 per day	1.0 to 1.5 kg/m^3 per day	See Tables 8.9, 8.12

Typical terms used to describe biofilter performance are based on either the volume of the media or its surface area. Although nitrification reaction rates are a surface area phenomenon, for fluidized sand beds and other granular media, the rate is easier expressed per unit volume rather than per unit surface area, due to the difficulty in measuring the actual media surface area. Table 7.5 provides a summary of design nitrification rates for general classifications of biofilters, e.g., warm or coolwater conditions and surface versus volumetric type medias. Chapter 8 provides specific design data for fluidized sand beds (see Table 8.12).

- **Volumetric** TAN conversion rate is the grams of TAN per volume per day converted into nitrate (g TAN/ft^3 per day or kg TAN/m^3 per day).

- **Areal** TAN conversion rate is the grams of TAN per unit surface area per day converted into nitrate (g TAN/ft^2 per day or g TAN/m^2 per day).

Malone and Pfeiffer (2006) established a "decision tree" that shows the numerous options available to system designers separated by the strategy used to provide oxygen and by the means of handling biofilm growth (Fig 7.4).

Figure 7.4 An organizational 'tree' of biofilters (Malone and Pfeiffer, 2006).

Suspended Growth or Fixed Film

The first juncture separates the two fundamental approaches to bacterial culture, suspended growth, or fixed film. Suspended growth systems were rarely found in production aquaculture until recently with the increased utilization of microbial floc systems for the production of very hardy species such as tilapia and marine shrimp. In these systems, heterotrophic bacterial growth is stimulated through the addition of organic carbonaceous substrate, for example molasses, sugar, wheat, cassava, etc. At high organic carbon to nitrogen feed ratios (greater than ~14 C/N ratios), heterotrophic bacteria assimilate ammonia-nitrogen

directly from the water replacing the need for an external fixed film biofilter (Avnimelech, 1999; McIntosh, 2001).

In the traditional intensive recirculating aquaculture production systems, large fixed-film bioreactors are used that rely on the nitrification of ammonia-nitrogen to nitrate-nitrogen by Ammonia Oxidizing Bacteria (AOB) and Nitrite Oxidizing Bacteria (NOB). In intensive recirculating systems, the growth of heterotrophic bacteria and the accumulation of organic carbon are minimized intentionally through the rapid removal of solids from the system and through water exchange. In general, fixed film bioreactors are more stable than suspended growth systems (Malone and Pfeiffer, 2006). In a fixed film biofilter, a thin bacterial biomass coats the filter media and the dissolved nutrients and oxygen are transported by diffusion into the biofilm. Numerous types of media have been employed to support this biofilm, including rock, shells, sand, plastic, and others too numerous to list. Just about anything that will support a biofilm and has a reasonable specific surface area (and many that do not) have been used over the years. The major drawback to these types of filters is that they can be quickly 'smothered' by heterotrophic bacteria, resulting in significant performance degradation. Malone and Pfeiffer (2006) subdivided fixed film biofilters into four fundamental blocks distinguished by the strategy used to provide oxygen and the techniques used to handle excess biofilm growth (Fig. 7.4).

EMERGENT BIOFILTERS

The second juncture separates fixed film biofilters based on the two fundamental methods of oxygen transfer. The 'emergent' filters use a cascading mixture of water and air over the media to insure a high level of dissolved oxygen at the surface of the biofilm. In the trickling filter, water is cascaded over the media in a tower opened to the ambient air. Whereas the rotating biological contactors create a similar effect by slowly rotating the media in and out of a tank of water, always keeping the media wet. Excessive biofilm is managed through the process of film sloughing or shedding, which demands a relatively high porosity to prevent clogging of the filter media with the sloughed film material. These filters provide a secondary benefit in the form of aeration and carbon dioxide stripping.

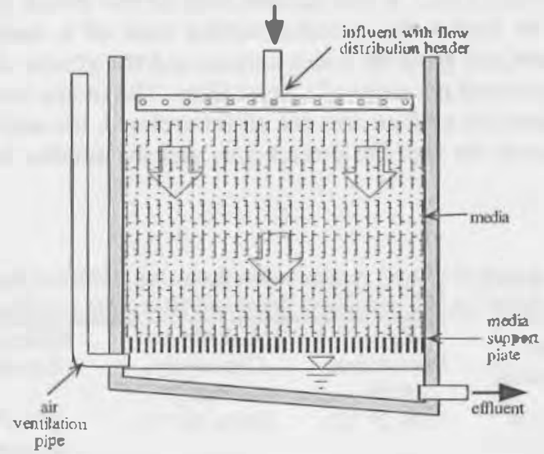

Figure 7.5 Schematic of a trickling filter with a rotary water distribution arm (from Summerfelt, 1999).

TRICKLING BIOFILTERS

Trickling filters consist of a fixed media bed through which a pre-filtered wastewater trickles down across the height of the filter. The wastewater flows downwards over a thin aerobic biofilm and dissolved substrates diffuse into the biofilm where they are consumed by the nitrifying bacteria. As it trickles over the media, the water is continuously oxygenated and carbon dioxide is removed by the ventilated air. Trickling filters have been widely used in aquaculture, because they are easy to construct and operate, are self-aerating and very effective at off gassing carbon dioxide, and have a moderate capital cost. Eding et al. (2006) has published an excellent review on the design and operation of trickling towers.

In municipal waste water treatment systems, trickling filters were traditionally constructed of rocks, but today most filters use plastic media, because of its low weight, high specific surface area (100–300 m^2/m^3) and high void ratio (>90%). According to Eding et al. (2006), Boller and Gudjer (1986) judged a specific surface area of 150–200 m^2/m^3 to be most suitable for the corrugated plastic media they applied in their ~~wastewater treatment research~~. Similar specific surface areas for plastic media (Bionet 160 m^2/m^3; Filterpac 200 m^2/m^3 and Munters 234 m^2/m^3) are installed in trickling filters applied in aquaculture (Kamstra et al., 1998).

A range of trickling filter design criteria has been reported. Typical design values for warm water systems are hydraulic loading rates of 100 to 250 m^3/day per m^2; media depth of 1–5 m; media specific surface area of 100–300 m^2/m^3; and TAN removal rates of 0.1 to 0.9 g m^2 per day surface area (note that this range of removal rate would have a 9X impact on the quantity of media required!). Trickling biofilters have not been used in large scale coldwater systems, due to the decrease in nitrification rates that occurs at the lower water temperatures and the relatively low specific surface area of the media. They have found a use in smaller hatchery systems where loads tend to be low and variable.

The trickling filter should allow space at the top for a water distribution device and should be open at the bottom to assure optimal ventilation. In some designs, the trickling filter is also used as a header tank for further distribution of water to the fish tanks and is closed at the bottom. In these designs, a blower has to be installed for forced ventilation (see Chapter 10 Gas Transfer for required gas to liquid airflow rates). Apart from nitrification and removal of BOD, a trickling filter is ideally suited for removal of carbon dioxide. Moreover, it can be used for evaporation cooling in warm climates. In both cases, a controlled airflow over the filter is needed. To realize this, the space on top of the trickling filter can be closed and connected to a ventilation system. For optimal degassing, the minimum ratio of air to water flow needed is in the order of 5-10 (see Chapter 10 Gas Transfer), while a minimum filter bed height is needed. When higher ventilation rates are applied, the increased evaporation may help in cooling the water during summer time. Forced ventilation also helps in preventing stagnant air in periods when the water temperature in the filter is almost similar to the air temperature outside the filter. Stagnant air reduces the oxygen partial pressure and results in poor aeration of the bulk water, which may subsequently reduce the nitrification capacity of the filter.

Eding et al. (2006) reported that the type of filter medium has an effect on the specific removal rate of ammonia (Kamstra et al., 1998). Cross flow media perform better than vertical flow or random flow media—an effect, which is attributed to, differences in hydraulic and wetting characteristics. Clogging of filter media can be a serious problem in commercial farms and must be avoided. In this respect, the effect of the hydraulic surface load of the filter and the type of filter material are difficult to quantify. Experience has shown that random flow media are prone to clogging, which is the reason why vertical flow and cross flow media have become more popular. Cross flow and vertical flow media come as self supporting blocks, which can be stacked easily and take out

when necessary. Random media are mostly in the form of loose 'balls' and require a special support frame.

A good water distribution device on top of the filter is essential to utilize the total filter volume (Fig. 7.6). Water can be distributed through a moving arm, a perforated screen, or a nozzle. In round filters with random media, a rotating beam is often applied. These constructions are sensitive to mechanical wear and need to be constructed carefully. Perforated screens are often used on small filters, but require frequent maintenance to avoid clogging of the holes. Nozzles (rotating) can handle large flows (Summerfelt et al., 2001) at little head pressure and can provide effective water distribution.

Figure 7.6 Two trickling filters with a rotary water distribution arm.

Rotating Biological Contactors (RBC)

A rotating biological contactor or biodisc filter, Fig. 7.7, is a fixed film bioreactor composed of circular plates aligned on a central axle, first developed for the treatment of treating domestic wastewater, (Van Gorder and Jug-Dujakovic, 2005; Brazil, 2006). The filter is usually staged in series within a flooded compartment through which recirculated water flows, with approximately half of the disc surfaces submerged, and half exposed to the air. The discs are rotated slowly (1.5 to 2.0 rpm) to alternately expose the biologically active media to the nutrient recirculated water and to the air, which provides oxygen to the biofilm. Early RBC designs were fabricated from discs of corrugated fiberglass roofing material. Currently, media with a higher specific surface area (258 m^2/m^3) are utilized in construction of RBC, which reduce the physical size and increase the ammonia and nitrite removal capacity. Hochheimer and Wheaton (1998) recommended a maximum hydraulic loading design

limit for RBCs of 300 m^3/m^2 day. Brazil (2006) determined an average total ammonia nitrogen areal removal rate of 0.43 ± 0.16 g/m^2 day for an industrial-scale, air-driven RBC used to rear tilapia at 28°C. Van Gorder and Jug-Dujakovic (2005) reported higher rates for multiple commercial scale systems of 1.2 g/m^2 day. In addition, carbon dioxide concentrations were reduced approximately 39% as the water flowed through the RBCs (Brazil, 2006) and 65% of the estimated carbon dioxide generated was off-gassed by the RBCs (Van Gorder and Jug-Dugakovic, 2005).

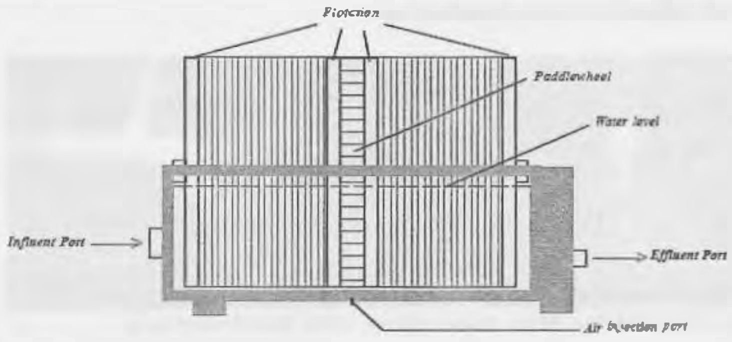

Figure 7.7 Schematic of a rotating biological contactor, RBC; axis of rotation is in same plane as the paper (from Van Gorder and Jug-Dujakovic, 2005).

Rotating biological contactors have inherent advantages for aquaculture, because they are self-aerating, require little hydraulic head, have low operating costs, provide gas stripping, and can maintain a consistently aerobic treatment environment. In addition, they tend to be self-cleaning due to the shearing of loose biofilm caused by the rotation of the media through the water. The main disadvantages of these systems are a) the mechanical nature of its operation, b) the substantial weight gain due to biomass loading of the media and the resultant load on the shaft and bearings, and c) the relatively high capital cost per unit of nitrification obtained (several fold higher than fluidized sand beds or microbead filters). Early efforts using RBC's often employed under-designed shafts and mechanical components, which resulted in mechanical failure, but a properly designed RBC is very functional and reliable. Figure 7.8 shows an RBC (manufactured by Fresh-Culture Systems, Inc.) categorized as "floating/air-driven/rotating biological contactor", which rotates using pumped air and/or water. Its weight is supported by the water column resulting in very little resistance to the rotation of the biofilter. This also eliminates the mechanical difficulties often reported with the gear motors, pillow blocks, chain drives and shafts used with many commercially available RBC's.

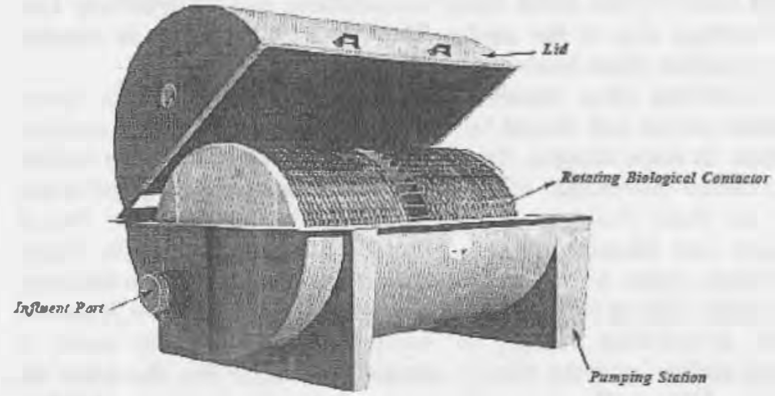

Figure 7.8 The Fresh-Culture Systems RBC 6000 rotating biological contactor (commercially available from Fresh-Culture Systems, Inc., 630 Independent Road, Breinigsville, Pennsylvania 18031).

SUBMERGED BIOFILTERS

The second major category of fixed film biofilters, submerged filters presume that sufficient oxygen can be transported to the biofilm in the water circulated through the filter. This is accomplished by the use of high recirculation rates, internal recycling, or through oxygen enrichment of the influent water (Malone and Pfeiffer, 2006). In addition, the assumption is made that ammonia diffusion into the biofilm is the rate limiting parameter and not dissolved oxygen. Thus, the goal of submerged filters is to first maximize the specific surface area in order to enhance nitrification. The three general types of submerged biofilters are categorized by the strategy used to manage biofilm accumulation (Fig. 7.4).

The first major category of submerged biofilters employ a fixed, static packed bed of media that has no active management of either the biofilm or solids accumulation. Examples of fixed, static packed beds are submerged rock biofilters, plastic packed beds and shell filters.

Submerged packed beds relay entirely upon endogenous respiration to control biofilm accumulation (Manthe et al., 1988). The water can flow either from the bottom up (upflow) or from the top down (downflow). Thus, the hydraulic retention time can be controlled by adjusting the water flow rate. Solids from the culture tank can accumulate within the submerged filter, along with cell mass from nitrifying and heterotrophic bacteria. This process can eventually block the void spaces, requiring some mechanism to flush solids from the filter for successful long term operation. To provide large void spaces to prevent clogging of the filters, the media used for submerged biofilters has been traditionally of large size, such as uniform crushed rock over 5 cm in diameter or plastic media over 2.5 cm in diameter. However, 5 cm diameter crushed rock would only have a specific surface area of 75 m^2/m^3 and a void fraction of only 40 to 50%. Random packed plastic media would also have a relatively low specific surface area of 100–200 m^2/m^3, but a much higher void fraction, greater than 95%. Drawbacks of this type of filter include problems of low dissolved oxygen and solids accumulation, resulting from heavy loading of organic matter (feed) and the difficulty of backflushing. Although this type of filter was promoted and used in aquaculture in the past, it has since been replaced in aquaculture due to the inherent high construction costs, biofouling problems, and operational expense. Packed submerged biofilters are still used in lightly loaded systems such as display aquaria and seafood-holding/display systems, where oyster shells are often used to help maintain calcium carbonate concentrations and other important trace minerals.

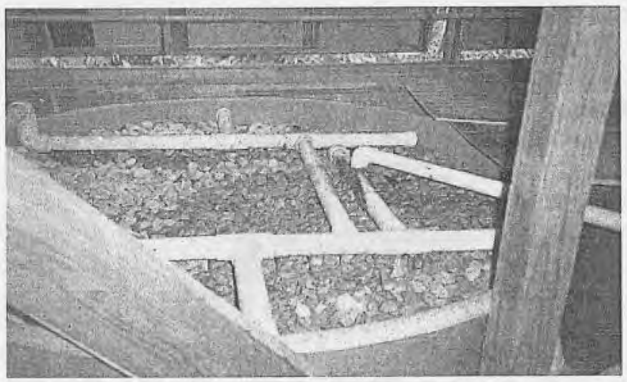

Figure 7.9 A submerged packed gravel biofilter with a pipe distribution manifold on the surface.

The second category of submerged biofilters utilizes a static bed that is intermittently "expandable" using air, water, or mechanical mixers. Excessive biofilm growth is removed by the process of abrasion as the media is agitated, then allowed to settle out before reintroducing the flow stream. Expandable biofilters are able to operate as mechanical filters for solids removal, biofilters for ammonia removal and as bioclarifiers accomplishing both solids capture and nitrification depending up design and backflushing frequency. Examples of expandable biofilters include upflow sand filters, floating bead bioclarifiers and foam filters.

PRESSURIZED UPFLOW SAND FILTERS

Still part of the second category of submerged biofilters, up flow pressurized sand filters or often a typical swimming pool filter are principally used as mechanical filters, although they may contribute some nitrification. They usually make for poor biofilters due to the high rate of backwashing and slow biofilm growth rates. Sand filters have been widely used for display aquaria. Up flow gravel filters have seen some utilization in large public aquariums, but are rarely used today because of the high water loss during backflushing. Very high flow rates are required through these biofilters to initiate their expansion.

FLOATING BEAD FILTERS

The floating bead filters are expandable granular filters that display a bioclarification behavior similar to sand filters (Fig. 7.10) (Malone and Beecher, 2000). They function as a physical filtration device or clarifier by removing solids (Chen et al. 1993), while simultaneously providing a large surface area for the attachment of nitrifying bacteria, which remove dissolved nitrogenous wastes from the water (Malone et al. 1993). Bead filters are often referred to as bioclarifiers for their ability to perform both biofiltration and clarification in a single unit.

Clarification is the process of removing suspended solids from the water. Suspended solids in aquaculture are generally small particles (< 100 micron) of undigested or partially digested food, bacteria, algae, clay, and silt, suspended in the water column. Bead filters remove the suspended solids by at least four different mechanisms as water is passed through the packed bed of plastic beads. Particles > 100 microns are subjected to physical straining. For slightly smaller particles (50 to 100 microns) the most dominant mechanism is settling. Suspended particles (5 to 50 microns) are removed by interception, a subtle process caused by collisions between the particle and the bead media surface. Finer

particles (< 20 microns) are removed through bioabsorption, the capture of particles by the bacterial biofilm.

Floating bead filters are resistant to biofouling and generally require little water for backwash. The bead filter is typically either bubble-washed or propeller-washed during its backwashing procedure, which expands the bed and separates trapped solids from the beads (Fig. 7.11). The beads used are food-grade polyethylene with a diameter of 3–5 mm and a specific gravity of 0.91, and a moderate specific surface area of 1150–1475 m^2/m^3 (Malone et al. 1993). Bead filters advantages include their modular and compact design, ease of installation, and operation. In addition, they can be used as a hybrid filter for both solids removal and nitrification.

The propeller-washed bioclarifiers are operated in the filtration mode most of the time. As recirculating water passes through the bed, suspended solids are captured and the biofiltration processes are active. Backwashing or cleaning of the bead bed is accomplished by turning off the pump and/or closing the inlet valve and then activating the mixing motor and propellers. The objective of the backwashing step is to release solids and excessive biofloc trapped between the beads. This is accomplished by the hydraulic shear forces induced by the propellers as the beads are thrust downward into the expansion zone and by contact between the beads as they swirl. The propeller washed bead filters are designed to input a lot of cleaning energy in a short period. Excessive washing just damages the biofiltration performance without benefiting clarification. Once the bed has been expanded and agitated for several seconds, the mixing motor is turned off and the settling mode of operation is initiated. Typically, the filter is left idle for 5 - 10 minutes. The beads float upward reforming the filtration bed, while the sludge is concentrated in the settling cone. The final mode of operation is sludge removal. Settling is very effective and it is not necessary to drain the filter completely. Commonly, the sludge drain line is equipped with a clear segment of pipe, which allows the clarity of the discharged water to be observed. As soon as the draining water appears to be as clear as the rearing tank's water, the sludge valve is closed. This approach greatly reduces water loss without affecting filter performance.

Another popular form of bead filter commonly used for small garden ponds and small aquaculture systems has an hourglass shaped internal geometry with a constricted washing throat. During continuous filtration, water from the production tank enters from the bottom through a slotted inlet pipe, flows upward through the bed of floating polyethylene beads and exits through a slotted discharge pipe at the top. The inlet pipe also serves as a sludge discharge line during backwashing. The discharge of

the filter is equipped with a valve (or check valve) that prevents the back-flow of air into the filter when the sludge (or drain) valve on the bottom is opened. This causes a vacuum to form within the filter housing. An air inlet valve, located on the side of the filter just below the washing throat, is opened so that air can be sucked into the filter as it drains. This constriction is critical because as the water leaves the filtration head, the beads are fluidized downward, and pass through the narrow throat where they are scrubbed further by the rising bubbles. The washing process is complete once the filter is drained and all the beads have dropped into the expansion chamber. Readjusting the valves and refilling the filter with the recirculation pump starts the next filtration cycle. In contrast to propeller-washed units, bubble-washed bead filters lose the entire water volume contained in the filter during backwashing. Both methods are easily automated with a simple controller. Bead filters using propeller-washed backflushing have been built with bead volumes of up to 2.8 m^3. Most small-scale systems use the bubble-washed filters, typically less than 0.28 m^3.

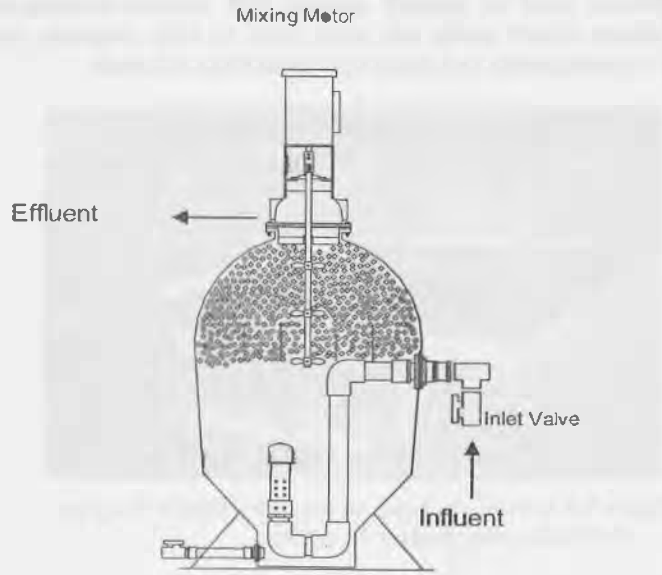

Figure 7.10 Schematic of a pressurized floating bead filter.

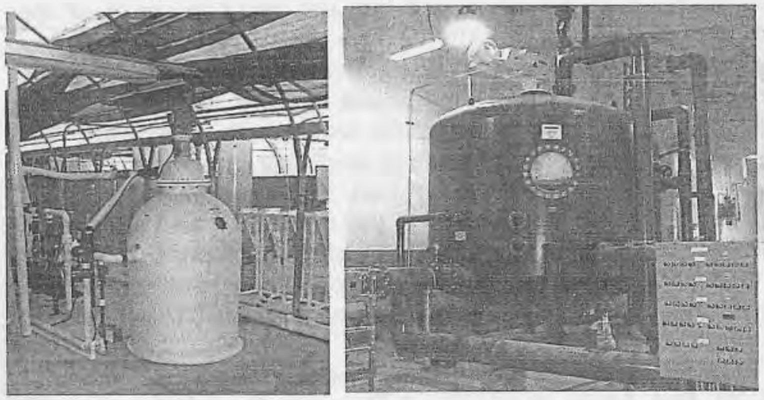

Figure 7.11 A PBF-10, propeller-washed and an on-site constructed commercial size propeller –washed bead filter used in a state hatchery.

The PolyGeyser Bead Filter is the next generation in Bead Filter technologies primarily through its automatic pneumatic backwash mechanism (Fig 7.12). Water is introduced below a bed of packed bead media and travels upward through the filtration chamber where mechanical and biological filtration takes place. Simultaneously, air is introduced into the air charge chamber at a constant, predetermined rate to achieve the desired backwash frequency. Once the charge chamber has reached capacity, the pneumatic trigger fires, releasing the entrained air from the charge chamber below the media bed. The sudden release of air from the charge chamber causes the beads to mix, roll, and "drop" as the air agitates the beads. The circulation pump/airlift operates continually, which ensures that the filter chamber begins refilling immediately after each backwash event. This causes the beads to float upward and reform as a bed. During the recharge cycle, suspended solids in the trapped backwash waters settle into the sludge storage chamber for later disposal via the sludge drain valve (usually every 2-3 days). At the same time, the supernatant is passed through the bead bed again as the air charge chamber is recharged with air.

The elimination of water loss associated with backwashing is a key element in this new filter. In most applications, dozens of backwash sequences can be automatically executed before sludge removal is required. There is no water loss associated with the backwash process and the water loss associated with sludge drainage is negligible. This strategy is particularly advantageous for marine systems, where the loss

of saltwater and need for large backwash water treatment units are minimized. The pneumatic strategy breaks the linkage between backwash frequency and water loss and allows the nitrification capacity of the unit to be fully utilized. Frequent backwash sequences have proven advantageous for optimizing the nitrification capacity of the unit. Numerous gentle scrubbing cycles promote high rates of nitrification by maintaining a healthy thin biofilm on the bead surfaces. Typical backwash cycles occur once every three to six hours. In recirculating bioclarifier applications, where the PolyGeyser Bead Filter operates concurrently as a clarifier and biofilter, total ammonia nitrogen (TAN) levels below 0.3, 0.5 and 1.0 mg-N/L can be expected at feed loading rates of 0.5, 1.0 and 1.5 pounds feed per cubic foot of EN bead media (8, 16 and 24 kg-feed/ m^3 day), respectively.

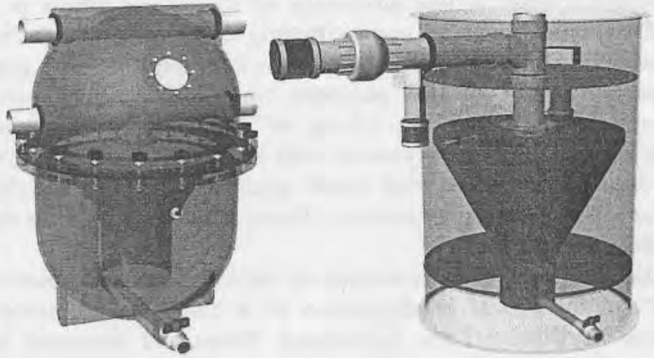

Figure 7.12 A 3 ft^3 and 25 ft^3 PolyGeyser Drop Filter (AST,LLC).

The third category of submerged biofilters, expanded bed, maintains the media in continuous expansion. These filters will no capture solids and the biofilm is continually abraded. These types of systems can use media with extremely high specific surface area, such very fine sands, or small plastic beads. Several examples of expanded bed biofilters are found in aquaculture including, fluidized-sand beds, microbead filters, and moving bed bioreactors.

MICROBEAD BIOFILTER

The microbead filter is distinctly different than the more commonly used floating bead filters (Timmons et al., 2006). Floating bead filters

work in pressured vessels and use a media that is only slightly buoyant. The required mass of beads for the volume required (~700 kg per cubic meter) make the media a relatively expensive component of a floating bead filter in contrast to sand or microbead media that is much less expensive on a per volume basis. Microbead filters use a polystyrene bead that is 1 to 3 mm in diameter compared to a floating bead filters media of approximately 3 mm in diameter. Microbeads are highly buoyant polystyrene beads with a bulk density of 16 kg/m^3. Beads are identified as Type A, B, C, or T with average diameters of 3, 2, 1.5 and 1 mm and specific surface areas of 1260, 1890, 2520, and 3936 m^2/m^3 respectively. Porosity of the media ranges from 36 to 40% with newer beads being closer to 40% and acclimated beads being 36% (Greiner and Timmons, 1998).

Microbead filters are considered a low-cost design alternative similar to fluidized sand filters because of their ability to be scaled to large production systems, also. A key advantage of microbead filters is that their cost of operation will be approximately 50% of a conventional fluidized sand bed due to the ability to use low head high volume pumps for their operation. For design purposes, microbead filters can be assumed to nitrify approximately 1.2 kg of TAN per cubic meter of media per day for warm water systems with influent ammonia-nitrogen levels from 2 to 3 mg/L. For cool water applications, rates should be assumed to be 50% of warm water rates. These rates are similar to those used for fluidized sand beds.

A microbead filter is a combination of trickling and granular type biological filters. A typical configuration of a small scale microbead filter is given in Figure 7.13. Microbead filters are operated in a downflow configuration where influent water is distributed over the top of the media bed and the water then trickles down through the media and flows by gravity out of the reactor vessel. The beads are the same material that is used for disposable drinking cups. Beads are created by a steam heat treatment of a raw crystal polymer. Trade name of the bead material is Dylite™ and can be obtained from distributors of Nova Chemicals Corporation, (Calgary, Alberta Canada). Once a local user is identified, beads will cost around $4 US per kg of material. Originally, microbead filters used Type T beads (1 mm) and more recently, users seem to prefer a Type B or C. Type A should be avoided.

Variations on microbead filters will be the manner in which the water is distributed over the beads. In Fig. 7.13, a spray diffuser is shown. Other applications use flooded perforated plates and create a water head of a few centimeters on top to create the water distribution. Some applications actually use the orifice plate to "hold down" the beads

and force the beads to be submerged into the retention vessel of the biofilter. Other designs intentionally create a gas space between the top of the beads and the water spray so that gas stripping can be forced. The ultimate embellishment in this arrangement is where the gas space is ventilated at 3 to 10 times the hydraulic loading rate to provide CO_2 stripping. All approaches can be made to work, but limitations imposed by specific designs need to be recognized. For example, a traditional trickling filter will result in gas stripping due to the high void ratio of the media. Microbeads will not provide gas stripping unless it is via the process of distributing the water over the top of the bead surface and additional design features are incorporated to flush high CO_2 laden air from the space above the bead surface.

The recommended minimum hydraulic loading rate is 1290 m^3/m^2 d (22 gpm/ft^2). Bed depths in microbead filters are limited to around 50 cm. It is unclear why, but the limited depth requirement is probably related to eventual channeling of water flow as the water trickles through the bead column. As the bed depth is increased, the opportunity for water channeling increases. Hence, the need either to limit bed depth or to provide some type of additional mixing or stirring of the bed. The bead bed will not act like a fluidized sand bed in terms of seeing the media being well mixed. The bead bed almost appears to be a static mass, but when loaded at the higher loading rates, one will observe movement of beads from the upper portion of the bed into the lower portions. To some degree, the bead bed acts like a slowly eroding sand castle, where outer portions of the bead bed column finally fall away from the walls back into the main portion of the bed where they are then re-mixed with other beads. This constant yet slow turnover of beads in the bed is what provides the overall volumetric nitrification of roughly 1 kg TAN per day per cubic meter of bead volume.

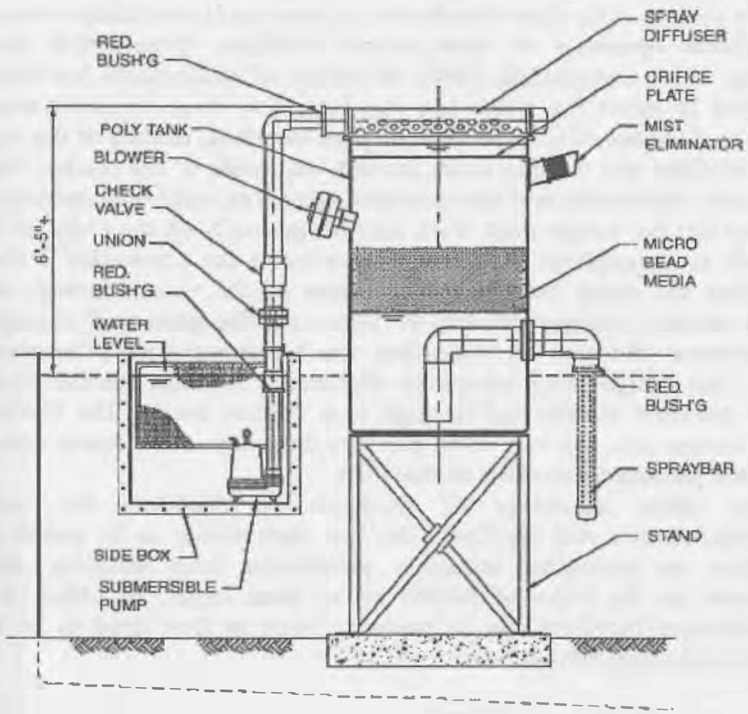

Figure 7.13 Generic microbead filter design (drawing courtesy of JLH Consulting, Courtenay, BC Canada).

Warm Water Application: Large scale applications of microbead filters using single reactor vessels have been implemented on large scale tilapia farms (in excess of 400 metric ton per year). Single microbead filter systems have been successfully operated for multiple years that assimilate the nitrogenous loading from a daily feeding of high protein feed (42%) of approximately 270 kg of feed per day distributed over a 24-hour period. For this particular application, the microbead filter had a bead depth of 21 cm, a filter bead volume of 6 m^3, and used a Type B bead 1.85 mm diameter. The hydraulic loading rate was 1108 $m^3/m^2/d$ (18.9 gpm/ft^2). Measured performance characteristics were: nitrification rate of 1.1 kg $TAN/m^3/d$, average influent TAN 1.9 mg/L, average system nitrite-N 0.6 mg/L, water temperature 26 C, and average feed per day into the system was 239 kg/d.

Coolwater Application: Microbead filters were applied to an Atlantic salmon smolt operation during the fall of 2004 using an average hydraulic loading rate of 984 $m^3/m^2/d$ (16.8 gpm/ft^2). Over a 30 day period, the average feeding rate was 56 kg (45% protein feed) for two biofilters (similar to Figure 7.13) with a media volume per biofilter of 0.93 m^3. Sodium bicarbonate was added daily at a rate of 13.5 kg/d, and makeup water averaged 567 Lpm (150 gpm). The average removal rate was 1.25 kg TAN per m^3/d with a removal efficiency per pass of 29% (SD 12%) across the filter. The water quality parameters in the fish tanks were TAN 1.2 (sd 0.9) mg/L, NO_2-N 1.4 (sd 0.3) mg/L, water temperature 13.6 C, hardness 72 mg/L, pH 7.3, turbidity 11.7 mg/L, CO_2 10 mg/L, and chloride concentrations 400 mg/L.

FLUIDIZED-SAND BIOFILTERS

Fluidized-bed biofilters have been used in several large-scale commercial aquaculture systems (15 m^3/min to 150 m^3/min or 400 to 4,000 gpm) (Figs. 7.14 and 7.15). Their chief advantage is the very high specific surface area of the media, usually graded sand or very small plastic beads. Specific surface areas range from 4,000 to 45,000 m^2/m^3 for sand versus 100 to 800 m^2/m^3 for trickling biofilter media and 1050 m^2/m^3 for bead filter media. The fluidized-bed biofilter can easily be scaled to large sizes, and are relatively inexpensive to construct per unit treatment capacity (Summerfelt and Wade, 1997; Timmons, 2000). Since the capital cost of the biofilter is roughly proportional to its surface area, fluidized-bed biofilters are very cost competitive and are relatively small in size compared to other types of biofilters (Summerfelt, 1999). Fluidized-bed biofilters are efficient at removing ammonia; typically removing 50–90% of the ammonia during each pass in cold- and cool-water aquaculture systems (Summerfelt et al. 2001). Nitrification rates for coldwater systems range from 0.2 to 0.4 kg TAN removal per day per cubic meter of expanded bed volume (Timmons and Summerfelt, 1998). In warmwater systems, TAN removal rates range from 0.6 to 1.0 kg per day per cubic meter expanded bed volume (Timmons et al., 1998). The main disadvantages of fluidized-bed biofilters are the high cost of pumping water through the biofilter and that a fluidized-bed biofilter does not aerate the water, as do trickling towers and RBC's. Additional disadvantages are that they can be more difficult to operate and can have serious maintenance problems, usually due to poor suspended solids control and biofouling.

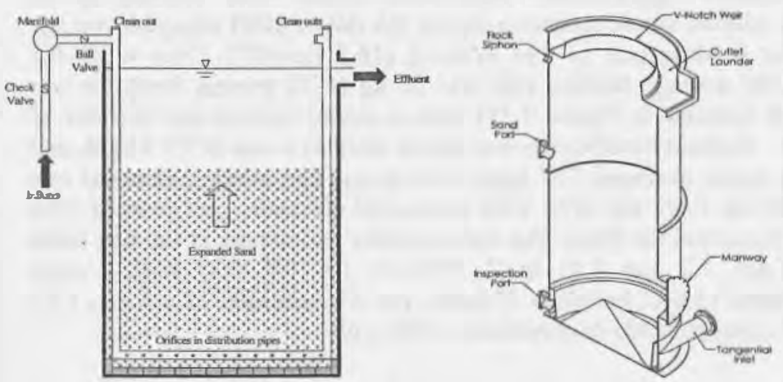

Figure 7.14 Schematic of two fluidized beds, one with a traditional pipe manifold (left) for water distribution and a Cyclo-Bio™ (right, drawing courtesy of HE Products, PO Box 145, Shandon OH 45063, 513-738-4333).

In fluidized-beds, water flows through the void spaces in the media, either upward or downward, depending upon the specific gravity of the media. The bed becomes fluidized when the velocity of the water through the bed is sufficiently large to suspend the media in the velocity stream, causing the bed to expand in volume. The resulting turbulent motion of the media provides excellent transport of dissolved oxygen, ammonia-nitrogen and nitrite-nitrogen to the biofilm and shears off excess biofilm. The result is high nitrification capacity in a relatively compact unit, but at the cost of the high energy required to fluidize the filter media.

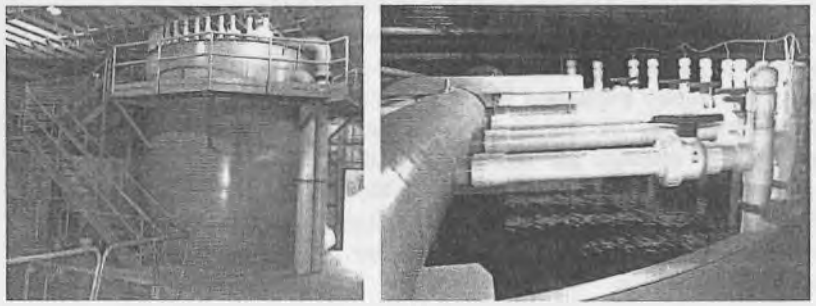

Figure 7.15 A 4 m diameter fluidized-sand biofilter and part of the pipe manifold distribution system being shown (right).

The design of the flow distribution mechanism is absolutely critical for reliable operation of fluidized-bed biofilters (Summerfelt and Cleasby, 1996; Summerfelt, 1996). A variety of mechanisms has been employed to inject the water into the bottom of large fluidized-sand biofilters. Traditionally, some form of pipe manifold, starting at the top of the biofilter and running down through the inside of the reactor, has been used. This header and lateral system creates an additional operating pressure that the pumps must work against, generally on the order of ⅓ to ½ of an atmosphere. A recent innovation is the Cyclo-Bio™ that introduces the water flow into the bottom of the vessel through an outside annulus (incorporated into the bioreactor fiberglass wall) through a continuous slot that circumscribes the bioreactor wall (Timmons, 2000). This design saves energy by eliminating the conventional pipe-lateral manifold system and its high pipe friction losses. The Cyclo-Bio™ design also has very little pressure drop due to the lower water velocities produced across the slotted inlet.

The major advantage of fluidized-sand biofilters for both conventional units and the Cyclo-Bio™ is their ability to be scaled to capacities to assimilate ammonia production from standing fish biomasses on the order of 50,000 kg or even larger. In effect, the fluidized-sand biofilters can be made as large as they need to be to handle a specified fish biomass.

MOVING BED BIOREACTOR (MBBR)

The moving bed bioreactor (MBBR) was developed in Norway in the early 1980's to reduce nitrogen discharge from municipal waste treatment plants into the North Sea (Figure 7.16). A significant advantage in upgrading existing waste water treatment plants was its small footprint and low maintenance in comparison to the operational and maintenance issues associated with trickling filters and rotating biological contactors. MBBR technology is currently widely used in European waste water treatment facilities and in both small and large scale commercial aquaculture operations.

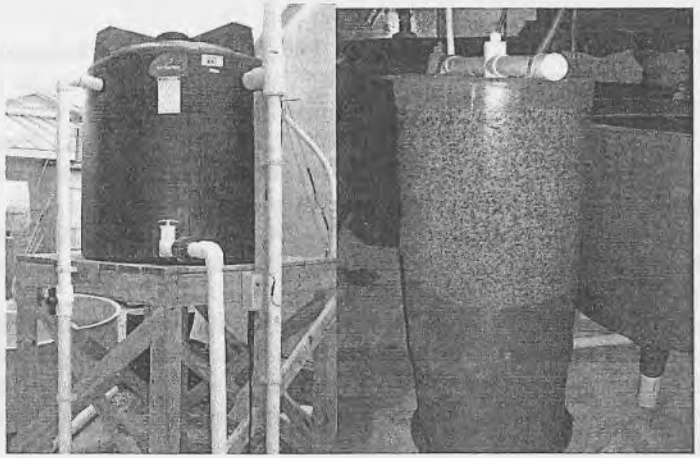

Figure 7.16 Two types of moving med bioreactors (MBBR's).

The MBBR is an attached growth biological treatment process based on a continuously operating, non-clogging biofilm reactor with low head loss, a high specific biofilm surface area, and no requirement for backwashing. The bacterial biomass grows on the media carriers and moves freely in the water volume of the reactor. The reactor can be operated under either aerobic conditions for nitrification or anoxic conditions for denitrification. For nitrification, the media is maintained in constant circulation via a course air bubble aeration system creating aerobic conditions and for denitrification via a submerged mixer for anoxic conditions.

Media usually occupies up to 70% of the reactor volume (normally 50% fill), in that at higher percentage fill reduces mixing efficiency. The media is kept within the reactor volume by a) an outlet sieve or screen, which may be vertically mounted, b) rectangular mesh sieves, or c) cylindrical bar sieves, vertically or horizontally mounted. The media most often used (Kaldnes K1) is made of high density polyethylene (density 0.95 g/cm^3) and shaped as a small cylinder with a cross on the inside of the cylinder and 'fins' on the outside (Ødegaard et al., 2004). Other media has also been used, although all have the characteristic of a protected area for biofilm growth.

Agitation within the reactor maintains the media in constant motion creating a scrubbing effect that prevents clogging and sloughs off excess biomass. Since MBBR's are an attached growth process, treatment capacity is a function of the specific surface area of the media. This is often reported as the specific surface area of the reactor, equal to the total

surface area of the media divided by the volume of the reactor, or the media specific surface area multiplied by the fraction of the total reactor volume that the media occupies. In some cases, the total surface area of the media that is available for biofilm development divided by the volume of the reactor is used, reflecting the significant abrasion of biofilm off the outer surface of some media types. For Kaldnes K1 media, the specific biofilm surface area is 500 m^2/m^3 and at 50% fill: 250 m^2/m^3 and at 70% fill: 350 m^2/m^3.

A model for predicting nitrification rates in MBBRs was developed by Rusten et al. (1995). For TAN as the rate limiting substrate (i.e. normally for most aquaculture systems), the following equation described the nitrification rate:

$$r_N = k(S_N)^n \qquad (7.12)$$

where r_N = nitrification rate, g TAN/m^3-day
k = reaction rate constant (1.3)
S_N = TAN concentration in the reactor, mg-N/L
n = reaction order constant (0.7)

A reaction order constant of n=0.7 was established by Hem et al. (1994) and the reaction rate constant (k) will depend upon the wastewater characteristics, temperature and other parameters that influence the growth of nitrifying organisms (Rusten et al., 2006). Figure 7.17 shows the nitrification rate as a function of substrate TAN concentration at 24 °C based on data from Rusten et al. (1995). For aquaculture systems, MBBR nitrification rates per m^3 of media on the order of 200 g/m^3-day for broodstock (<0.3 mg-N/L), 400 g/m^{3}day for fingerling (<0.5 mg-N/L) and 800 g/m^{3}day for growout (<1.0 mg-N/L) can be expected.

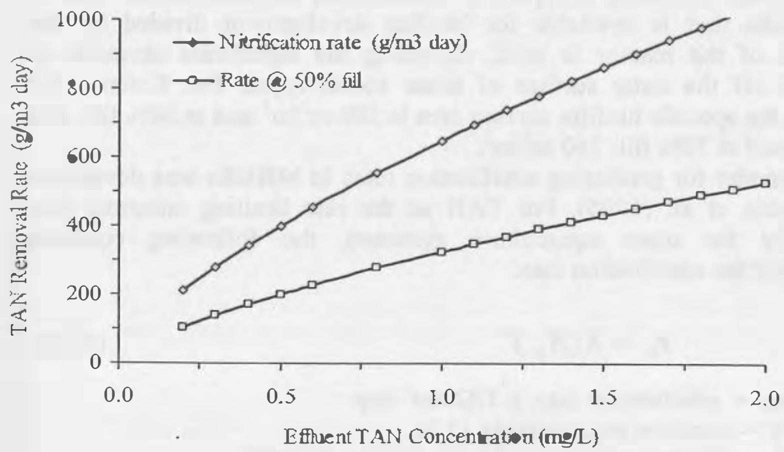

Figure 7.17 Influence of TAN concentrations on TAN removal in a Kaldnes MBBR at 24 °C. Based on nitrification rate equation (Eq. 7.12) and data from Rusten et al. (1995).

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CHAPTER 8
BIOFILTER DESIGN

8.0 INTRODUCTION

Designing a biofilter for an aquaculture system is a complicated balancing act in which the designer needs to make numerous choices and compromises in order to find an optimum solution or at least one that works given the numerous constraints. The final goal of any design is to strike a balance between the initial capital costs, operating costs, and risk management, while optimizing productivity and profitability. This starts at the initiation of the project when species and overall production goals are specified. Design criteria such as system volume, culture density, feed rates, temperature and many others--constrain the biofilter design. The key at this stage is to set realistic values for each of these constraints and to provide a sufficient, but not excessive safety factor. Too large a safety factor results in unnecessary high capital and operating costs. Too low a safety factor will limit system ability to respond to ever changing biomass and nutrient loads on a system and unexpected system 'upsets'. In addition, compounding of many small safety factors needs to avoided, which can result in the unintentional exaggeration of system design.

Manufacturers of biofilters would like to see some form of standardized criteria for sizing and comparing different commercially available biofilters (Drennan, et al., 2006). In addition, a standardized labeling system would allow system designers to accurately and rapidly determine which biofilter(s) meet their needs. To this end, the development of standardized rating and design procedures for biological filters (Colt, et al., 2006) has been initiated through the Standards and Reporting Committee of the Aquaculture Engineering Society (AES). For all research reports, detailed information on the physical size of the filter, media characteristics (type, density, size, and specific surface area), water flow rates, and specific loading rates will be required. The filter performance will be evaluated at several total ammonia nitrogen concentrations under standardized conditions (temperature, salinity, carbon/TAN or BOD/TAN concentrations, dissolved oxygen, and alkalinity). It should be acknowledged that biofilter performance studies are difficult to conduct due to the large number of parameters that must be controlled and the number of measurements that must be completed. Yet to facilitate accurate design and comparison of biofilters and

systems, this research is important to minimize selection based on personal experience, marketing promises, and other nontechnical criteria.

8.1 Generalized Engineering Considerations

All of the biological filters reviewed in Chapter 7 are designed to perform the same function: oxidizing ammonia and nitrite to nitrate. Thus, the biological filter must be designed to fully oxidize the ammonia-nitrogen produced, with an additional safety margin to account for unforeseen events. From a practical perspective, the biofilter selection is less critical in small production systems, i.e., systems that feed at rates below 50 kg per day, than for larger production systems. In small systems, biofilters can be over-designed and the added cost is generally not of critical importance to the overall economic success of the venture. Smaller operations are not competing in the wholesale market where margins are extremely small and can make a producer non-competitive due to high capitalization costs. Usually, the smaller growers target niche markets and gain sales by providing service or other product attributes that allows premium pricing to the seller/grower. On the other hand, for large production systems, i.e., those feeding 100 kg or more per day, economical designs with both low capital cost and low operating cost for the biological filter are much more critical.

One of the more difficult design parameters to estimate is the oxygen demand of both the species being grown and the biofilter. Using stoichiometry, the oxygen demand could be as low as 0.37 kg of oxygen per kg of feed fed (0.25 for fish metabolism and 0.12 for nitrification). For design purposes however, using a ratio of 1.0 kg oxygen per kg of feed is a good starting point. It must be recognized that in small production systems, the efficiency of use will often be poor for a wide variety of reasons, e.g., lack of qualified personnel, lack of attention to maintenance, leakage. Of course, these things could happen in a large farm, too. It is also essentially impossible to go to the theoretical lower limit, as systems will always have varying degrees of suspended solids and heterotrophic activity and thus additional oxygen demand. The best most efficient oxygen usage in a commercial operation of any scale in the authors experience is around 0.5 kg of oxygen use per kg of feed.

"Rule of Thumb"

For design purposes, using a ratio of 1.0 kg oxygen per kg of feed is a good starting point

Each biofilter described in Chapter 7 has advantages and disadvantages that need to be considered during the early design phase. One of the chief advantages of both the trickling biofilter, MBBR and the RBC is that they add oxygen to the water flow during normal operation. In addition, they provide some carbon dioxide stripping. In contrast, the submerged biofilters, bead filters, and fluidized-bed biofilters are all net oxygen consumers and rely completely on the oxygen in the influent flow to maintain aerobic conditions for the biofilm. If for whatever reason, the influent flow is low in dissolved oxygen; anaerobic conditions are generated within the biofilters.

Both the trickling biofilters and the RBC have the disadvantage of having low specific surface area media. Since the capital cost of the filter is proportional to its total surface area, the result is physically large and more costly filters. In contrast, bead filters and especially fluidized-bed filters use media with a high specific surface area, which results in reduced cost and space requirements in comparison to that required to achieve the same surface area in a trickling biofilter or MBBR.

An additional disadvantage of some trickling biofilters and RBC's is that they readily biofoul, if suspended solids are not adequately controlled. Carbon eating heterotrophic bacteria grow significantly faster than the autotrophic nitrifiers do. Their mass can double in an hour, while it takes nitrifiers days to double. This high growth rate and the associated oxygen demand consequently suffocate the nitrifiers buried deeper in the biofilms, resulting in death and sloughing of the biofilm from the bioreactor surfaces. The advantages and disadvantages of several types of biofilters are summarized in Table 8.1 (Wilton, 2001).

Before moving on to the next section on design, we would like to emphasize one critical point that affects all biofilter performance. You must have effective solids removal BEFORE the high ammonia water is transferred (pumped) to the biofilter. As mentioned above, the heterotrophic growth will compromise the nitrifiers ability to oxidize ammonia, mostly because the heterotrophs consume the oxygen prior to the oxygen being able to diffuse into the biofilm to where the nitrifiers are. This results in the nitrifiers being starved for oxygen and then they die-off resulting in complete sloughing of the biofilm and loss of nitrification capacity. A recipe for disaster in a RAS is to have poor solids removal. Actively plan for effective solids removal prior to the water being moved into the biofilter.

> **"Rule of Thumb"**
>
> A recipe for disaster is to have poor solids removal; this causes biofilter failure!

8.2 DESIGN PARAMETERS: WHERE DO YOU START?

The design of any biofilter requires at minimum the physical size of the filter, media characteristics (type, density, size and specific surface area), water flow rates and the filter performance (Colt et al., 2006). We previously gave the reader a table in Chapter 7 that describes ammonia assimilation rates for two general classifications of biofilters (dense media such as sands or non-dense such as used in trickling filters, see Table 7.5).

To develop standardized rating and design procedures for biological filters, filter performance must be evaluated and reported in a standardized manner. To this end, Colt, et al. (2006) reported results of a preliminary study of standards for biofilter performance studies. Colt emphasized the importance that critical parameters be defined and reported in a standard manner, both in terms of definition, variable names, and units. Depending on the type and scale of an experiment, reporting of certain parameters will be either mandatory or optional. The results of this study are summarized in Tables 8.2, 8.3, 8.4, 8.5 and 8.6, corresponding to the general categories of media characteristics, filter characteristics, general system influent characteristics, biofilter performance, and system performance. Not all of these design parameters are necessary for basic biofilter design, but many are useful in comparing alternative design scenarios.

Table 8.1 Advantages and Disadvantages of Commonly Used Biofilters (Wilton, 2001).

Pro's	Con's
Trickling Biofilters	
Very simple design and construction requirements	Some biofilm sheared off is large enough to be problematic and many systems integrate post-biofiltration mechanical filtration for this reason
Currently a very popular method of biofiltration in the wastewater industry, which should improve material availability and cost	Filters using this media type tend to be very large in high feed load coldwater systems
Allows for passive aeration and CO_2 removal concurrent with biofiltration	Media itself can be costly due to low specific surface area
Media and design assistance is currently available from reputable commercial vendors facilitating the design effort	
Systems using these types of filters tend to be extremely stable	
Rotating Biological Contactors (RBC's)	
Low energy to move fluid across media	Can be expensive due to low specific surface area for large scale facilities
Provides passive aeration for nitrification process and limited CO_2 control	Mechanically more complex than most other biofilters
Can allow for efficient facility layout and combination of several processes (mechanical filtration, biofiltration, aeration and pumping in one common sump)	Subject to rotational wear on bearing surfaces
Amenable to modularization, which can be useful for development of scalable facilities	

Pro's	Con's
Bead Filters	
Well developed product available from reputable commercial vendors. Can simplify system design and construction	Can be expensive due to relative low specific surface area for large scale facilities
Can be combined with other filter types in interesting hybrid systems as alternative design method	Relatively high head loss across filter can be an operational cost consideration
Can in some cases improve fine particle removal rates in well designed systems	Variable head loss across system can be problematic in systems without variable speed pumps
Amenable to modularization, which can be useful for development of scalable facilities	Has potential to leach nutrients into system or to fuel heterotrophic bacteria growth if not installed with pre-filtration systems or is back flushed infrequently
Fluidized-Bed Biofilters	
Very economical to build from commercially available materials	Can have problems with media carryover (initial fines) on system start-up
Large amount of design effort specific to coldwater systems using these types of filters	There are historical anecdotal reports of intermittent bed motility and system crashes
Raw filter media has very high specific surface area at low cost, which allows for very conservative design allowing for inherent capacity for expansion or load fluctuation	Can have problems with restarting if not designed to account for bed re-fluidization and distribution manifold/lateral flushing
Widest installed base of coldwater biofilters offers large operational and design experience base to draw from	Media density changes over time with biofilm accumulation in fine sand filters typical of coldwater systems, which necessitates a bed growth management strategy
Can be field built using a variety of proven methods or purchased from established and reputable vendors opening many design and construction options for facility designers or operators	Some systems can require relatively expensive plumbing to ensure that media is not back-siphoned on pump shut-down or power failure

Table 8.2 Important Design Characteristics or Parameters for Biofilter (not all parameters apply to all filters); Colt, et al., 2006

Parameter	Basis or source of parameter	Units	Symbol
Filter height	Measured; floor to top of tank	cm or m	H_{tank}
Water height	Measured	cm or m	H_{water}
Water discharge height	Measured with respect to grade	m	$H_{discharge}$
Filter volume—no media	Computed	m^3 or L	V_0
Filter Cross-sectional area	Computed	m^2	A_{cross}
Total active surface area	Computed	m^2	A_{media}
Media volume	Computed	m^3	V_{media}
Reactor volume	Computed	m^3	$V_{reactor}$
Hydraulic loading rate	$1.44Q_{filter}/A_{cross}$	$m^3/(m^2\ d)$	L_{hyd}
Hydraulic media loading rate	$1.44Q_{filter}/A_{media}$	$m^3/(m^2\ d)$	L_{media}
Bed height—no flow	Measured	cm or m	BH_0
Bed height—operating	Measured	cm or m	BH_{op}
Water distribution system	Manufacturer or supplier	information	
Submergence (RBC)	Measured	%	D
Rotational speed (RBC)	Measured	rpm	ω
Biofilter flow	Measured or calculated	lpm	Q_{filter}
Make-up flow	Measured or calculated	lpm	Q_{mu}
Rearing unit flow	Measured or calculated	lpm	Q_{ru}
Reuse flow	Measured or calculated	lpm	Q_{reuse}
Discharged flow	Measured or calculated	lpm	Q_{out}

Table 8.3 Important Design Characteristics or Parameters for Media (Colt et al., 2006)

Parameter	Basis or source of parameter	Units	Symbol
Manufacturer	Manufacturer or supplier	Information	
Type	Manufacturer or supplier	Information	
Nominal size	Manufacturer or supplier	mm or cm	
Material	Manufacturer or supplier	Information	
Media dimensions	Manufacturer or supplier	cm or cm x cm	
Specific surface area	Manufacturer, supplier, or measurement	m^2/m^3	SSA
Specific gravity media	Manufacturer or supplier	Value	SG

Table 8.4 Important Design Characteristics or Parameters for General Influent of Culture System Supply (Colt, et al., 2006)

Parameter	Basis or source of parameter	Units	Symbol
Culture species	Operator	Information	
Feed rate and type of feed	Estimated	kg/d and information	FR
Feeding frequency	Operator	#/d	
Protein content of feed (as fed)	Nitrogen x 6.25	%	
Cumulative feed burden	$10^6FR/1440Q_{mu}$	mg/L (or ppm)	CFB
Cumulative oxygen consumption		mg/L	COC
Cumulative loading	kg fish/Q_{ru}	kg/lpm	CL
Total ammonia nitrogen	Measured	mg/L as N	TAN_{in}
Nitrite-nitrogen	Measured	mg/L as N	NO_2-N_{in}
Nitrate-nitrogen	Measured	mg/L as N	NO_3-N_{in}
5-d biochemical oxygen demand	Measured	mg/L	BOD5
Chemical oxygen demand or total organic carbon	Measured	mg/L	COD_{in} or TOC_{in}
Alkalinity	Measured	mg/L as $CaCO_3$	ALK_{in}
pH	Measured	pH units	pH_{in}
Dissolved oxygen	Measured	mg/L	DO_{in}
Carbon dioxide	Measured	mg/L	CO_{2in}

Table 8.5 Important Design Characteristics or Parameters for Filter Performance (Colt, et al., 2006)

Parameter	Basis or source of parameter	Units	Symbol
Temperature	Measured	C	T
Total ammonia $nitrogen_{out}$	Measured	mg/L as N	TAN_{out}
Nitrite-$nitrogen_{out}$	Measured	mg/L as N	NO_2-N_{out}
Nitrate-$nitrogen_{out}$	Measured	mg/L as N	NO_3-N_{out}
Percent TAN removal	$(TAN_{in} - TAN_{out})/TAN_{in}$	%	PTR
Filter system ratio	$1440\ Q_{filter}(TAN_{in} - TAN_{out})/$ (total TAN removed d)	%	FSR
ΔO_2	$DO_{out} - DO_{in}$	mg/L	ΔO_2
ΔCO_2	$CO_{2out} - CO_{2in}$	mg/L	ΔCO_2
ΔpH	$pH_{out} - pH_{in}$	mg/L	ΔpH
Nitrite-nitrogen generation	$(NO_2\text{-}N_{out} - NO_2\text{-}N_{in})/TAN_{in}$	%	NO_{2gen}
Nitrate-nitrogen generation	$(NO_3\text{-}N_{out} - NO_3\text{-}N_{in})/TAN_{in}$	%	NO_{3gen}
Volumetric TAN conversion rate	$1440Q_{filter}(TAN_{in} - TAN_{out})/V_{media}$	mg TAN/(m^3 d)	VTR
Volumetric nitrite conversion rate	$VTR + VNR_A$	mg NO_2-N/(m^3 d)	VNR
Surface TAN conversion rate	$1440Qf_{filter}(TAN_{in} - TAN_{out})/A_{media}$	mg TAN/(m^2 d)	STA
Volumetric oxygen consumption rate	$1440Q_{filter}(TAN_{in} - TANout)/V_{media}$	mg oxygen/(m^3 d)	$VOCR_{tot}$
Volumetric oxygen consumption rate for nitrifying bacteria	(3.47VTR + 1.09VNR)(0.92)	mg oxygen/(m^3 d)	$VOCR_{nit}$
Volumetric oxygen consumption rate for heterotrophic bacteria	$VOCR_{tot} - VOCR_{nit}$	mg oxygen/(m^3 d)	$VOCR_{het}$
Oxygen consumption ratio	$VOCR_{nit}/VOCR_{tot}$	%	OCR

Table 8.5 (cont.) Important Design Characteristics or Parameters for Filter Performance (Colt, et al., 2006)

Parameter	Basis or source of parameter	Units	Symbol
Pumping power (average daily)	Computed using actual filter head losses, bed height, and 70% efficiency	kW	P_{pump}
Other power (average daily)	Measured or estimated	kW	P_{other}
Total power (average daily)	Computed from above parameters	kW	P_{tot}
Ammonia removal efficiency	$1440Q_{filter}(TAN_{in} - TAN_{out})/24P_{tot}$	mg TAN/kWh	ARE
Ammonia removal efficiency (system)		mg TAN/kWh	ARE_{sys}
Oxygen utilization efficiency	$(1440Q_{filter}\,\Delta DO)/24P_{tot}$	mg O_2/kWh	OUE

Table 8.6 Important System Performance of Biofilter – Culture System (Colt, et al., 2006).

Parameter	Basis or source of parameter	Units	Symbol
Volumetric feed capacity	(kg feed/d)/V_{media}	kg feed/(m^3 d)	VFC
Volumetric biomass capacity	kg biomass/V_{media}	kg biomass/m^3	VBC
Volumetric feed capacity efficiency	$VFC/24P_{tot}$	kg feed(m^3 kWh)	VFCE
Volumetric biomass capacity efficiency	$VBC/24P_{tot}$	kg biomass(m^3 kWh)	VBCE
Loop strength	106(kg/d feed)/$1440Q_{ru}$	mg/L (or ppm)	LS

8.3 DESIGN EXAMPLE: BIOFILTRATION

The following biofilter design examples continue the engineering design for the construction and operation of an Omega Fish Aquaculture Facility. Omega Industries (OI) would like several options for biofiltration systems for the production of 45.4 metric tonnes per year of Omega Fish (Scientific Name: ***Physhi physhy***). Tables 3.4 and 5.14 are included here as summaries of what has been determined to this point (see Chapters 3,4 and 5, **Tables** 3.4 and 5.14 for original development and discussion).

Table 5.14 Summary of Design Total Volume and Design Flows for the Two Design Scenarios

	Design Scenario One		Design Scenario Two	
	Two Juvenile or Fry Pods	Five Fingerling/ Growout Pods	Single Juvenile Pod	Two Fingerling/ Growout Pods
Pod Total Volume:	32.0 m^3 (8,465 gal)	57.2 m^3 (15,110 gal)	96.1 m^3 (25,400 gal)	143.8 m^3 (38,000 gal)
Total Flow:	90.8 m^3/hr (400 gpm)	90.8 m^3/hr (400 gpm)	273 m^3/hr (1200 gpm)	227 m^3/hr (1000 gpm)
Center Discharge:	22.7 m^3/hr (100 gpm)	22.7 m^3/hr (100 gpm)	68.3 m^3/hr (300 gpm)	45.4 m^3/hr (200 gpm)
Side-wall Discharge:	68.1 m^3/hr (300 gpm)	68.1 m^3/hr (300 gpm).	205 m^3/hr (900 gpm)	181.7 m^3/hr (800 gpm)
Feed Rate Per day:	10.9 kg (24lbs)	27.5 kg (60.5 lbs)	20.9 kg (46 lbs)	63.5 kg (140 lbs)

The first step in the design process is to calculate the dissolved oxygen requirement for both the fish and the biofilter. In this example, the oxygen demand has been estimated at 0.50 kg DO/kg feed. As previously mentioned, this is often a difficult number to determine, and must either be based on existing system performance or estimated from literature data. As discussed in Chapter 4, as a starting point when lacking any research or engineering data, fish metabolism is often estimated at 250 g O_2/kg feed. For a trickling tower, RBC and MBBR,

the ambient atmosphere will supply the oxygen requirement for nitrification and for any heterotrophic bacteria that is attached to the filter or in the water column. At the very minimum, the overall oxygen demand could consist of only the fish metabolism of the feed. But in the real world, there is always additional oxygen demand from bacterial, so we increase the design value to of 0.5 kg O_2/kg feed as a minimum. For submerged biofilters both the fish metabolism and the biofilter demand must be satisfied from the influent water to the biofilter making a practical design value of 1.0 kg O_2/kg feed.

Table 3.4 Initial and Final Weights and Lengths of the Three Stage Omega Fish Production Strategy

	Initial Wt & Size	Final Wt & Size	Final Tank Biomass per tank	Feed Rate (%bw/day)	Final Feed Rate per day
Fry or juvelile:	50 g	165 g	193 kg	1.56%	3.0 kg
	13.4 cm	19.9 cm		(1.1 FCR)	
Fingerling:	165 g 19.9 cm	386 g 26.4 cm	450 kg	1.28% (1.2 FCR)	5.7 kg
Growout:	386 g 26.4 cm	750 g 32.9 cm	875 kg	1.11% (1.3 FCR)	9.6 kg

The following design examples are for the fingerling/growout stage of production with five separate Pods on five LSS (Table 5.14, Design Scenario One). Each Pod consists of two fingerling tanks and two growout tanks stocking at 5 week intervals. This reduces the maximum biomass and feed loading on the LSS. Thus the maximum biomass per Pod will be around 2,267 kg (5,000 lbs) and the maximum feed rate per day about 27.5 kg (60 lbs).

8.4 Design Example: Trickling Tower

For dimensioning or sizing a trickling filter, only limited information is available (Eding et al., 2006). In practice, TAN removal efficiency is often empirically determined for a fixed set of successful conditions such as fish species, feed load, filter height, filter media type, hydraulic surface load, suspended solids unit, and TAN influent concentration. TAN removal rates can also be determined from empirical relationships

(Liao and Mayo, 1974) and from the kinetics of TAN removal (Bovendeur et al., 1987; Heinsbroek and Kamstra, 1990).

The simplest case is when the TAN removal efficiency for a certain trickling filter influent concentration is known, based on data for a fixed filter height, media type, hydraulic surface load, TAN removal rate, and temperature. The required total nitrification surface area (A_{media}, m^2) is calculated from the trickling filter TAN load (P_{TAN}, kg/day) and the estimated nitrification rate (r_{TAN}, g TAN/m^2/day). The bioreactor volume (V_{media}, m^3) is a function of the total filter surface area (A_{media}, m^2) and the specific surface area (SSA, m^2/m^3 biofilter media) of the filter media. The shape of the reactor depends on the hydraulic surface load (HLR, m^3/m^2/ day) (Losordo et al., 2000; Wheaton et al., 1995).

The design of a trickling tower follows a simple set of steps with the nitrification rate based on the active surface area of the media and since the trickling tower is self aerating, the oxygen requirement is limited to fish metabolism plus some allowance for bacteria consumption. Note that the filter type will partially dictate the design value used for oxygen demand. In this example, we will use 0.50 kg of oxygen per kg of feed fed, since a trickling filter provides sufficient oxygen for the nitrifiers on the trickling media (we increase the fish metabolism oxygen demand of 0.25 to 0.50 kg/kg feed to allow for oxygen demand in the water column by heterotrophic bacteria). Note that as a "rule of thumb", we recommend using 1.0 kg of oxygen per kg of feed fed. Be very cautious in choosing this design value.

Step 1: Calculate the water quality loads on the system (oxygen, CO_2, TAN, and TSS).
Step 2: Calculate controlling water flow requirement (Q_{tank}).
Step 3: Calculate TAN production by fish (P_{TAN}).
Step 4: Calculate the surface area of media (A_{media}) required to remove P_{TAN} from the areal TAN removal rate (ATR).
Step 5: Calculate the volume of media required (V_{media}) based upon the required area and the specific surface area (SSA) associated with the media being used.
Step 6: Calculate the biofilter cross-sectional area ($A_{biofilter}$).
Step 7: Calculate the biofilter depth from the biofilter cross-sectional area ($A_{biofilter}$) and volume (V_{media}).

The first biofilter option choice investigated for one of the fingerling and growout pods is utilizing a trickling tower biofilter whose media has a SSA of 200 m^2/m^3 (61 ft^2/ft^3). The water flow requirements are based choosing the highest of the calculated flow rates for each of the

control variables (oxygen, CO_2, TAN, TSS, and tank exchange). The biofilter sizing is based on the daily TAN production rate, which is directly proportional to the feeding rate of 27.5 kg/day. Metric units will be used throughout this example.

Both Step 1 and 2 were demonstrated in Chapter 3 to calculate the controlling flow rate for the maintaining DO, TAN, and CO_2 (see Table 3.5). Here, we need to re-do these calculations since our feeding load is now for a pod (27.5 kg feed/day) as opposed to an individual tank. Following the example from Chapter 3 and using the same design parameters for efficiency and tank water quality target values, we arrive at a set of required flow rates for the pod, Table 8.7.

Table 8.7 Required Flow Rates for a Pod (Scenario One, Table 5.14) with a Daily Feed Load of 27.5 kg feed/day

Parameter	C_{best} (mg/L)	Treat Removal	C_{tank} (mg/L)	C_{out} (mg/L)	Load (kg/day)	$Q_{required}$ (m^3/hr)
Oxygen	16	90%	5	14.90	13.75	57.9
CO_2	0.5	60%	20	8.90	18.91	67.3
TAN	0	35%	2	1.30	.885	52.6
TSS	0	90%	10	1.00	6.88	31.8

This is a very good example of how the additional design constraint of tank water exchange rate can influence the design. In this particular case, each of the four tanks (two fingerling and two growout tanks) on the pod required approximately 378 Lpm (100 gpm) of water flow to promote effective hydraulics in the fish tank, resulting in an overall flow rate for the pod of 1514 Lpm (90.8 m^3/hr or 400 gpm).

Table 3.5 Required Flow Rates for Oxygen, TAN, Carbon Dioxide, TSS and Tank Exchange (*controlling flow rate)

Water Quality Parameter	Juvenile/Fry	Fingerling	Growout
Oxygen*	321 Lpm (85 gpm)	360 Lpm (95 gpm)	337 Lpm (89 gpm)
TAN	95 Lpm (25 gpm)	185 Lpm (49 gpm)	310 Lpm (82 gpm)
Carbon Dioxide	121 Lpm (32 gpm)	208 Lpm (55gpm)	333 Lpm (88 gpm)
TSS	68 Lpm (18 gpm)	132 Lpm (35 gpm)	189 Lpm (50 gpm)
Tank Exchange	321 Lpm (85 gpm) (20 min HRT)	374 Lpm (99 gpm) (30 min HRT)	390 Lpm (103 gpm) (45 min HRT)

Step 3: Calculate TAN production by fish (P_{TAN}) assuming the feed has a protein content of 35%, and as shown in Table 5.14, at a design feeding load of 27.5 kg of feed per day..

$$P_{TAN} = a_{TAN} * R_{feed}$$

$$= (0.092 * 0.35) \frac{kg_{TAN}}{kg_{feed}} * 27.5 \frac{kg_{feed}}{day}$$

$$= 0.885 \frac{kg_{TAN}}{day}$$

Step 4: Calculate the surface area (A_{media}) required to remove P_{TAN} from the Areal TAN removal rate (ATR). Based on our experience with trickling towers (also, see Table 7.5 for range of values that can be used), the estimated Areal TAN removal rate is 0.45 g TAN/m^2 day.

$$A_{media} = \frac{P_{TAN}}{ATR}$$
$$= \frac{0.885 \dfrac{kg_{TAN}}{day} * \dfrac{1{,}000 g}{kg}}{0.45 \dfrac{g_{TAN}}{m^2 day}} = 1970\, m^2$$
$$1970\, m^2 \cdot \frac{10.76 ft^2}{m^2} = 21{,}200\, ft^2$$

Note that for coldwater applications (12–15°C) when TAN concentrations entering a trickling column are less than 1–2 mg/L, then the Areal TAN removal rate is only about 0.15–0.25 g TAN/m^2 day. Note also that the Areal TAN removal rate drops to only 0.1–0.2 g TAN/m^2 day for saltwater applications (@ 24°C) when TAN concentrations entering the trickling filter are 1–2 mg/L (although there are mixed data on these estimates, assuming a 30% reduction for saltwater applications is a guideline)..

Step 5: Calculate volume of media based on the specific surface area (SSA) associated with the media being used, for example, BioBlock has a SSA of 200 m^2/m^3 (61 ft^2/ft^3).

$$V_{media} = \frac{A_{media}}{SSA}$$
$$= \frac{1970\, m^2}{200 \dfrac{m^2}{m^3}} = 9.83\, m^3 \text{ or } 347\, ft^3$$

Step 6: Calculate the biofilter cross-sectional area from required flow for the fish oxygen demand (Q_{tank} = 90.8 m^3/hr or 1514 Lpm) and the design hydraulic loading rate; HLR of 255 m^3/m^2 day (4.4 gpm/ft^2) required to prevent clogging of the media bed (note that higher hydraulic loading rates can be used and this value is considered a minimum value to create effective wetting of all surfaces).

$$A_{bed} = \frac{Q_{tank}}{HLR}$$
$$= 1{,}514 \frac{L}{min} * \frac{1\, m^3}{1{,}000 L} * \frac{1}{255 \dfrac{m^3}{m^2}} * \frac{1{,}440\, min}{day}$$
$$= 8.55\, m^2 \text{ or } 92 ft^2$$

The diameter of a single biofilter tank, $D_{biofilter}$, with this cross sectional area is:

$$D_{biofilter} = \sqrt{\frac{4 * A_{bed}}{\pi}} = \sqrt{\frac{4 * 8.55\, m^2}{3.14}} = 3.30\, m \text{ or } 10.8 ft$$

In this case to keep the filter size relative small, two or more filters might be used, so the cross sectional area of each of these biofilter units would be (8.55/2 = 4.28 m^2). The diameter would be:

$$D_{biofilter} = \sqrt{\frac{4 * A_{bed}}{\pi}} = \sqrt{\frac{4 * 4.28\, m^2}{3.14}} = 2.33\, m = 7.7\, ft$$

Step 7: Calculate the biofilter depth ($Depth_{media}$) from the biofilter cross-sectional area (A_{media}) and volume (V_{media}).

$$Depth_{media} = \frac{V_{media}}{A_{media}} = \frac{9.83\, m^3}{8.55\, m^2} = 1.15\, m = 3.8 ft$$

Apart from nitrification, a trickling filter is ideally suited for removal of carbon dioxide (Eding et al., 2006). Moreover, it can be used for evaporation cooling in warm climates. It is critically important with trickling towers to remove as much of the suspended solids to prevent clogging of the media. To accomplish this, rotating drum filters with a mesh size of 30 to 60 µm are often used ahead of the trickling tower (Eding et al., 2006).

8.5 DESIGN EXAMPLE: RBC

For dimensioning or sizing a rotating biological contactor, only limited information is available (Van Gorder and Jug-Dujakovic, 2005,

Brazil, 2006). In practice, TAN removal efficiency is often empirically determined for a fixed set of successful conditions such as fish species, feed load, specific filter design, and TAN influent concentration. The simplest case is when the TAN removal efficiency for a certain RBC filter is known, based on performance data.

The design of a RBC follows the same set of steps as the trickling tower with the nitrification rate based on the active surface area of the media and since the RBC is also self aerating, the oxygen requirement is limited to fish metabolism. Using the initial design parameters of the trickling tower design example, the design process is the same up to Step 4.

Step 4: Calculate the surface area (A_{media}) required to remove P_{TAN} from the Areal TAN removal rate (ATR).

Based on research with several commercial scale RBC systems, Van Gorder and Jug-Dujakovic (2005) determined an Areal TAN removal rate of 1.2 g TAN/m^2 day. This was for a commercial scale RBC Model 10000, 1.22 m in diameter with a surface area of 930 m^2. Thus, each RBC Model 10000 was able to remove approximately 1.13 kg TAN/day. Approximately one tank volume per hour flowed through the biofilters and the system TAN concentration averaged 3 mg/L at 26 °C. This would suggest that one of these RBC's would be sufficient to handle the ammonia load for this design example of 0.885 kg TAN/day. In addition to nitrification, significant amount of carbon dioxide would be off-gassed by the RBC.

8.6 Design Example – Floating Bead Biofilter

The primary method for sizing the bead volume of floating bead filters is based on an organic loading rate (Malone and Beecher, 2000), assuming that the biofilter is used both as a bioclarifier to remove organic solids and as a filter to sustain nitrification. Since essentially all organics in a recirculating system is from the feed, the sizing criterion, v_f is based on the feed load, Table 8.7. The volume of bead media required for a specific application, V_{media} (m^3), can be determined from the design load, R_{feed} (kg/day), and the floating bead filter sizing criterion, v_f (kg feed/m^3 of media·day):

$$V_{media} = R_{feed} \cdot v_f \tag{8.1}$$

The feed load criterion, v_f, of 16 kg/m^3 media day has been tested and proven to be stable in the local commercial sector (Beecher et al. 1997; DeLosReyes et al. 1997; Sastry et al. 1999). At this design criterion, the floating bead filters can reliably provide solids capture, BOD reduction, and nitrification, while sustaining water quality conditions suitable for the growout of most food fish species, i.e., TAN and nitrite-nitrogen levels can be expected to remain well below 1 mg/L (Malone and Beecher, 2000). Reduction of the sizing criterion to 8 kg/m^3 media day allows the reliable maintenance of water quality conditions demanded by the juvenile and fingerling category. Finally, a loading guideline of 4 kg/m^3 media day is recommended for breeding and broodstock maintenance programs where the cost of maintaining pristine water quality conditions is justified by the value of the stock.

Table 8.8 Performance Parameters for Pressurized Bead Filters (Malone and Beecher, 2000)

Performance Parameter	Units	Growout Systems	Fingerling Systems	Broodstock Systems
Feed loading, v_f	kg feed/m^3 media·day	≤16	≤8	≤4
Design TAN concentration	mg-N/L	1.0	0.5	0.3
Typical TAN concentration	mg-N/L	<0.5	<0.3	<0.1
Volumetric TAN conversion rate	g TAN/m^3 media·day	140–350	70–180	35–105
Enhanced Nitrification Media	g TAN/m^3 media·day	210–530	105–270	50–157
Volumetric oxygen consumption rate	kg O_2/m^3 media·day	2.5–3.0	1.4–2.5	0.7–2.5
Temperature	°C	20–30	20–30	20–30
Filter effluent dissolved oxygen	mg/L	>3.0	>3.0	>3.0
Alkalinity	mg/L as $CaCO_3$	>100	>80	>50
pH	pH units	7.0–8.0	6.8–8.0	6.5–8.0
Aggressive backwash interval	days	1–2	1–3	1–7
Gentle backwash interval	days	0.5–1	1–2	1–3

An alternative approach to sizing floating bead filters is based on the areal nitrification capacity (Malone et al., 1993). This criterion is based on the observation that for a wide range of floating bead filters, their areal conversion rates (ATR) are approximately 300 mg TAN/m^2 day, in systems with TAN and nitrite levels between 0.5 and 1.0 mg N/L (Malone and Beecher, 2000).

Observed VTR values tend to increase with the increasing TAN tolerances (0.3, 0.5 and 1.0) associated with the three categories in Table 8.9. These VTR values can then be used to estimate the size of the floating bead filter:

$$V_{media} = (1.0 - I_s)\frac{R_{TAN}}{VTR} \qquad (8.2)$$

The in situ nitrification fraction (I_s) in Eq. 8.2 recognizes the effect of nitrification occurring in the water column, on the sidewalls of the tank, and in the system piping configuration (Mia, 1996). This fraction is often conservatively estimated at 30%, although values in excess of 50% have been observed. The authors of this text ignore this contribution in favor of over designing the system to insure long term stability. Also, this effect seems less noticeable as systems are scaled to large commercial applications (e.g., 500 ton per year and larger systems), Additional information and detailed design criteria can be found in Malone and Beecher (2000).

Continuing the example of the design process for the Omega fish facility, the water flow requirements are based on the tank water exchange requirements and the sizing of the floating bead filter is based on the daily TAN production rate, which is directly dependent on the feeding rate. Metric units will be used throughout. So, the first 3 steps of the design process remain the same (i.e., calculating P_{TAN}).

Step 4: Calculate the volume (V_{media}) required to remove P_{TAN} from the Volumetric TAN removal rate (VTR) (see Table 8.9). Assume a growout application, and a VTR at the maximum TAN value of about 1 mg/L, 350 g N/ m^3 day.

$$V_{media} = (1 - I_s)\frac{P_{TAN}}{VTR}$$

$$= (1.0 - 0.0) * \frac{0.885 \frac{kg_{TAN}}{day} \cdot \frac{1{,}000 g}{kg}}{\frac{350 g_{TAN}}{m^3 day}} = 2.53 m^3 \ or\ 89.2\ ft^3$$

This design assumes an in situ nitrification fraction of 0.0, which is the conservative approach which produces the largest media requirement. A 1.42 m^3 (50 ft^3) propeller washed bead filter is commercial available, constructed of heavy-duty food grade fiberglass. Design flow rates are from 760–1140 Lpm (200–300 gpm; see www.beadfilters.com for design specifications on these filters). Two of these biofilters would be sufficient for this design, incorporating additional safety factors by having two separate filters and increasing the flow through the system and thus the overall oxygen carrying capacity of the system. This could be critical during periods of overloading of the system, increased oxygen demand due to unforeseen events, or if one biofilter is taken off-line.

The new PolyGeyser Bead Filter sizing criteria are highly dependent on the application in which the filter is utilized. For aquaculture applications, where the bead filter acts as a bioclarifier, providing both solids capture and biological filtration, these filters are sized according to the maximum amount of feed (35% protein dry pellets) that is fed per day. Where the PolyGeyser Bead Filter will be used as a clarifier, the filter is sized either according to the maximum daily feed input or by the required flow rate to be compatible with the biological filter. Table 8.9 summarizes the filtration capacities for PolyGeyser Bead Filters for several applications. The criteria presented in this table already have a substantial safety factor included. The bioclarifier category is divided into three sub-categories reflecting the changes in water quality objectives. For aquaculture fingerling/ornamental, growout two separate loading guidelines are provided for warm water and cold water conditions. In both cases, the criteria are designed to ensure that a TAN level below 0.5 mg-N/L can be achieved. These are peak sustainable loading guidelines, meaning that a filter can sustain the indicated TAN concentration at the peak loading for an indefinite period. Finally, a set of criteria is provided for broodstock and fry systems that provide very pristine water quality with a maximum TAN below 0.3 mg-N/L.

Table 8.9 PolyGeyser Bead Filter sizing criteria. (Aquaculture Systems Technologies, 2006)

Filter Model	DF-3	DF-6	DF-10	DF15	DF-25	DF-50
Media Volume (ft^3)	3	6	10	15	25	50
Surface Area (ft^2)	1,200	2,400	4,000	6,000	10,000	20,000
Flow Rate (gpm)	45	90	150	225	375	750
Grow-out*	4.5	9.0	15	22.5	37.5	75
Fingerling Grow-out* @ > 15 °C	3	6.0	10	15	25	50
Fingerling Grow-out* @ < 15 °C	2.25	4.5	7.5	11.25	18.75	37.5
Broodstock/Fry/Holding/ Conditioning*	1.5	3.0	5.0	7.5	12.5	25.0
Solids Only (lbs feed/day)	15	30	50	75	125	250

* Max Loading – Lbs 35% Protein Feed/ day

As a design example, consider using a PolyGeyser Bead Filter for biofiltration of the fingerling/growout pod where 27.5 kg (60 lbs) of feed are fed each day. Based on Table 8.9, two DF-15 PolyGeyser filters in warm water (>15 °C) would be able to handle as much as 30 kg (66 lbs) of feed per day, providing an additional small safely factor. The allowable maximum flow rate for this filter is 850 Lpm (225 gpm), which is less than the designed requirement for fish tank water exchange (Q_{tank} = 90.8 m^3/hr or 1514 Lpm or 400 gpm, but higher than the flow required for oxygen control),. So either you would have to upsize to two DF-25's or re-evaluate your tank water exchange rates, particularly for the growout tanks, e.g., use a longer HRT than 45 minutes. Increasing your allowable HRT design could negatively impact the ability of a tank to effectively remove solids from the center drain.

8.7 Basic Design Concepts: Fluidized-Bed Sand Biofilter

All biological filters are designed to perform the same finction of oxidizing ammonia and nitrite to the fully oxidized form of nitrate. From a practical perspective, biofilter selection is less critical in small

production systems, but the right choice is essential for economical reasons in large operations. One primary benefit of a fluidized-bed biofilter is the ability to scale the biofilter to the system needs without paying large economic penalties as the fluidized-bed biofilter is scaled to a larger size. It is conceivable that a several hundred ton per year facility might be operated on one to three biofilters for the entire facility.

Continuing the example of the design process for the Omega fish facility utilizing a fluidized-bed biofilter. As previously shown, the production of total ammonia nitrogen (TAN) is approximately 3.2% by weight of the fish feed (35% protein) being fed per day, or more specifically 0.092% of the protein fed per day. Therefore, if 27.5 kg of feed are fed per day to the growout tank system, then there will be approximately 0.89 kg per day of TAN produced.

Biofilter TAN Removal Rates and Efficiencies

The biological filter must be designed to fully oxidize this TAN production; otherwise, the TAN concentrations in the tank will rise above design levels. Table 8.10 provides design values for TAN removal for both cool and warmwater systems and as affected by sand diameter size. This data is based upon replicated 10 cm (Cornell University) and 15 cm (Freshwater Institute) fluidized-bed biofilters. The sands were fluidized at fixed velocities that were set to achieve bed expansions of 50% with clean sand, e.g., a bed that is 1 meter in static depth would be 1.5 meters in depth when expanded. At Cornell University, inlet concentrations were 0.6 to 0.7 mg/L TAN, temperature 26°C, pH 7.3, 6 to 7 mg/L DO, and TSS <10 mg/L. At the Freshwater Institute, inlet concentrations were 0.5 to 0.6 mg/L TAN, temperature 15°C, pH 7.3 to 7.5, 10 to 11 mg/L DO, nitrite 0.04 to 0.06 mg/L, and TSS <10 mg/L (Tsukuda et al. 1997).

Note that the TAN removal rates in Table 8.10 are expressed on a unit volume basis, instead of on a surface area basis. As previously mentioned, low density medias such as RBC's and trickling towers provide nitrification rates proportionate to surface area provided by the media, but current research suggests that nitrification rates in granular medias are much more closely related to volume of media than surface area provided by the media. The large surface area provided by small sands provides no advantage in terms of nitrification rate. The fine sand biofilters, however, have demonstrated a much higher TAN removal efficiency (90% removal), when compared to the large sand biofilters (10–17%). The high removal efficiencies found with the finer sands are in part due to the low velocities required to fluidize fine sands. The lower velocities result in longer hydraulic retention times (HRT) in the reactor

vessel and an associated higher percentage removal of ammonia. The negative aspect is that the reactor volume must be proportionately larger than a reactor that uses a lower HRT and larger diameter sands.

Table 8.10 Average TAN Removal Rates and Efficiencies Measured Across Coldwater (15°C) and Warmwater (26°C) Biofilters as a Function of Sand Size

Sand retained between sieve mesh sizes	40/70	20/40	18/30
Coldwater (15°C) Systems (Tsukuda et al. 1997)			
TAN removal rate, kg/d/m^3 clean static bed	1.5	0.51	0.51
TAN removal rate, kg/d/m^3 expanded bed	0.41	0.35	0.35
TAN removal efficiency, % each pass	90	10	10
Warmwater (25°C) Systems (M. B. Timmons, unpublished data)			
TAN removal rate, kg/d/m^3 clean static bed	*NR	~1	~1
TAN removal rate, kg/d/m^3 expanded bed	NR		
TAN removal efficiency, % each pass	NR	10 to 20	5 to 10

*NR = not recommended due to excessive biofilm growth

Based upon the data in Table 8.10, it seems reasonable to use a nitrification rate of 1.0 kg TAN/m^3 per day of static bed as a design value for warmwater systems and a rate of 0.7 kg TAN/m^3 per day for clean static sand for coldwater systems (15°C).

The most important factors in the design of a biofilter are (1) the mass of TAN that it removes per day, i.e., the product of the flow rate across the biofilter and the change in concentration of ammonia across the biofilter; and, (2) the TAN removal efficiency (f_{rem}) of the biofilter. The mass of TAN removed per day can often be increased as the hydraulic loading rate is increased across a biofilter. However, increased hydraulic loading rate can decrease the TAN removal efficiency as an increased hydraulic loading rate shorten the water retention time within the biofilter and increases the mass load on the filter.

The f_{rem} of TAN controls the accumulation of TAN in the recirculating system and thus the concentration of TAN discharged from a culture tank (TAN_{out}, mg/L). The TAN_{out} can be estimated using Eq. 8.3, which was first developed by Liao and Mayo (1972):

$$TAN_{out} = \left\{\frac{1}{1 - R_{flow} + \left(R_{flow} \cdot f_{rem}\right)}\right\} \cdot \left\{\frac{R_{TAN}}{Q} \cdot \frac{m^3}{1000\,L} \cdot \frac{10^3\,mg}{1\,g} \cdot \frac{\min}{60\,s} \cdot \frac{1\,day}{1{,}440\,\min}\right\} \tag{8.3}$$

where R_{TAN} (g TAN per day) is the rate at which the waste is produced, R_{flow} is the fraction of water flow that is reused, and Q (m^3/s) is the flowrate of water recirculated through the biofilter. This equation was derived from mass balances that assume that no waste accumulation can occur in a culture tank, that the make-up water contains no TAN, and that the recirculating system is operating under steady-state conditions, i.e., water flow rates, waste production rates, and unit process treatment efficiencies are relatively constant.

The primary multipliers in Eq. 8.3 actually represent the "waste accumulation factor due to reuse" and the "concentration of waste that would be produced in a single pass through the tank." Therefore, Eq. 8.4 can be re-written as:

$$TAN_{out} = \left\{\begin{matrix}\text{accumulation factor}\\ \text{due to reuse}\end{matrix}\right\} \cdot \left\{\begin{matrix}\text{concentration of waste}\\ \text{produced in a single pass}\\ \text{through the culture tank}\end{matrix}\right\} \tag{8.4}$$

where,

$$\left\{\begin{matrix}\text{accumulation factor}\\ \text{due to reuse}\end{matrix}\right\} = \left\{\frac{1}{1 - R_{flow} + \left(R_{flow} \cdot f_{rem}\right)}\right\} \tag{8.5}$$

and

$$\left\{\begin{matrix}\text{concentration of waste}\\ \text{produced in a single pass}\\ \text{through the culture tank}\end{matrix}\right\} = \left\{\frac{R_{TAN}}{Q} \cdot \frac{m^3}{1000L} \cdot \frac{10^3\,mg}{1\,g} \cdot \frac{min}{60\,s} \cdot \frac{1\,day}{1{,}440\,min}\right\} \tag{8.6}$$

According to Eq. 8.3, the accumulation of waste within a recirculating system is dependent upon R_{flow} and the f_{rem} across the unit treatment process, i.e., the biofilter in the case of TAN. Most RAS operating in temperate climates reuse high fractions of their flow (to

conserve heated water) and generally operate with only 5 to 100% of the total system water volume exchanged daily, equivalent to a fraction of water flow reused R ≥ 0.96. In such recirculating systems, waste accumulation depends mainly upon the f_{rem} across the water treatment units and Eq. 8.4 simplifies to:

$$TAN_{ss} \cong \left\{\frac{1}{f_{rem}}\right\}\cdot\left\{\frac{R_{TAN}}{Q}\cdot\frac{m^3}{1000L}\cdot\frac{10^3\ mg}{1g}\cdot\frac{\min}{60\,s}\cdot\frac{1\,day}{1{,}440\min}\right\} \quad (8.7)$$

Trout, char and salmon require relatively clean water and low levels of un-ionized ammonia, so high f_{rem} are required when designing systems to raise these species. For this reason, fluidized-sand biofilters containing fine sands are commonly used in coldwater recirculating systems because these biofilters will often achieve 70–90% TAN removal efficiencies. Fluidized-sand biofilters using fine sands are also capable of providing complete nitrification (due to their excess supply of surface area), which helps to maintain low nitrite-nitrogen concentrations within the recirculating system (generally <0.1–0.2 mg/L as nitrogen).

Species such as tilapia and African catfish do not require the high quality water required by salmonids. Therefore, the f_{rem} of TAN across the biofilter will not be as important as the mass of TAN that the biofilter removes each day.

Fine sands are not recommended for use in warmwater systems, due to probable difficulties in controlling excessive biosolids growth at warmer temperatures. Control of biosolids growth is still an issue when fine sands are used in coldwater applications, but biosolids growth is relatively easy to manage at 12–15°C.

Volume of Media Required

Given a defined rate of TAN production in g/d, and choosing an appropriate removal rate from Table 8.10, the volume of media required, V_{media}, can be determined:

$$V_{media} = \frac{P_{TAN}}{VTR} \quad (8.8)$$

As a general rule, design a fluidized sand bed with as much sand media volume as you can, i.e., minimize the ammonia loading on the bed per unit volume of sand media. This minimizes biological growth on the media and results in a less difficult bed to manage. The volume of sand

required is independent of any of the other calculations, since the TAN removal rate is considered to be independent of the velocity through the bed.

"Rule of Thumb"
For fluidized sand bed,
1.0 kg TAN/d/m³ warmwater
0.7 kg/TAN/d/m³ coolwater
(static sand bed volume)

Sand Bed Depth and Biofilter Cross Sectional Area

The selection of sand depth is primarily influenced by the physical constraints of the building, ceiling height, whether the sand filter can be partially submerged into the ground, and whether any additional elevation is required to gravity flow filtered water back to the culture tank through aeration and/or oxygenation unit processes. In any case, the design should result in requiring as much sand volume as practically possible. The larger the sand volume, the lower TAN loads per unit volume of sand, which provides a factor of safety for the overall design. It also minimizes the biological film growth per unit particle of sand, which in turn will minimize sand bed growth and management problems related to changing expanded sand bed depths.

When fine sands are used, growth of biofilm can increase the expanded bed depth to the point that sand will be flushed from the reactor into the rest of the system. Therefore, fine sand biofilters are usually designed for eventual expansion of the biofilm-coated sand bed to achieve 200 to 300% of initial static sand depth, e.g., if a clean sand depth is 1 m before fluidization, after 50% fluidization the clean sand will be 1.5 m, once a thick biofilm grows in this bed the total sand expansion may be 200%, or around 3 m of total depth.

Based upon the above guidelines, then, choose and define your unexpanded bed depth, h_{sand}. Then, given the sand bed depth and the volume of media required for ammonia removal, the cross sectional area of the sand bed can be calculated:

$$A_{bed} = \frac{V_{media}}{h_{sand}} \quad (8.9)$$

SELECTING SAND SIZE

The selection of the proper sand size is related to the previous discussion on sand depth. The overall design for the fish system will include calculating flow rates to maintain target levels for the various water quality parameters: primarily oxygen, ammonia, and carbon dioxide. Depending upon the type of oxygenation and CO_2 stripping units being used, large variations in required design flow rates can result. Thus, the design of the different components of the overall system is often manipulated until the flow rates for the different water quality parameters being controlled by the different system components are somewhat in balance.

Selecting the sand size and bed expansion determines the water velocity through the biofilter, V_{sand}. A finer sand will require lower water velocities than will a larger sand. Selecting smaller sands will result in larger reactor vessels and higher HRT's. The rate of TAN production by the fish will determine the pumping rate from the fish tank. This flow rate is the same flow rate that the biofilter vessel will see. Given a defined water flow rate, the selection of sand size will determine the allowable velocity through the biofilter vessel, V_{sand}, which can be expressed as gpm per ft^2 (m^3/day per m^2). The flow rate per unit area reduces to (unit balance):

$$\frac{m^3/day}{m^2} \cong \frac{m}{day} * \frac{100\,cm}{m} * \frac{day}{1440 \cdot 60s} \cong \frac{cm}{s} \tag{8.10}$$

BED EXPANSION AND THE ASSOCIATED REQUIRED FLOW RATE

The failure of the fluidized-bed biofilter to properly expand can result in severe problems. Under-fluidization of the fluidized-bed biofilter will result in bed channeling with water leaving the biofilter untreated. Degrees of poor fluidization will result in the larger sands moving to the bottom of the bed and becoming static. Such areas then are apt to become anaerobic or anoxic resulting in denitrification and other undesirable water chemistry changes, e.g., sulfide gas production.

The authors' recommendation is to choose an overall clean sand expansion of around 50% when designing the fluidized sand bed system. More generally, we would recommend expansions to be between 40 to 100%. Table 8.11 provides the required V_{sand} as affected by sand size and degree of expansion chosen. Larger expansions ensure greater assurance that all the sands are fluidized, but this comes at a higher flow rate or a reduced biofilter cross-sectional area. Choosing too low of an expansion will increase the likelihood that some of the largest sand particles will not fluidize and may create anaerobic pockets at the bottom of the sand bed column. Avoid this at all costs! Anaerobic pockets can lead to hydrogen sulfide gas (toxic at extremely low concentrations) and most certainly off-flavor conditions in the fish.

Sand expansion is a function of water temperature and of several sand characteristics. Water velocity requirements increase with increasing expansion and increasing sand size. Estimates of expansion velocity requirements for a given sand can be erroneous due to the influence of water temperature on viscosity, as well as by variations in sand characteristics from different quarries.

Construct hydraulic testing columns with careful attention to how the flow is distributed and use at least 1 m sand depth. Experiences with small test columns of 10 to 15 cm diameter have not been reliable in predicting full scale bed behavior. The observed fluidization and expansion in these smaller columns is very sensitive to the flow distribution mechanism. Try to use as large a diameter test column as possible and also try to mimic the flow distribution system that will be used on the biofilter units for the full-scale system.

The expansion water velocity values in Table 8.11 must be applied with caution because each particular sand source may have specific characteristics that cause it to expand more or less than the values predicted. We recommend that you develop your own data for your own specific sands and use the data in the Table 8.11 as an estimate or starting prediction point to design your sand filters.

Table 8.11 Upflow Velocity Requirements for Specific Sand Size and Degree of Expansion

Sieve Designation Number	D_{eq} (mm)	Specific Surface Area (m^2/m^3)	Velocity (cm/s^*) Estimated to Achieve Fluidization at: 0% exp (ε=0.45)	20% exp (ε=0.542)	50% exp (ε=0.633)	100% exp (ε=0.725)	150% exp (ε=0.780)
100	0.15	29,530	0.024	0.030	0.081	0.23	0.39
80	0.18	24,859	0.034	0.045	0.14	0.35	0.56
70	0.21	20,952	0.048	0.086	0.23	0.51	0.77
60	0.25	17,600	0.068	0.15	0.35	0.71	1.02
50	0.30	14,815	0.10	0.24	0.51	0.95	1.32
45	0.35	12,429	0.14	0.36	0.71	1.24	1.69
40	0.42	10,476	0.19	0.51	0.94	1.58	2.09
35	0.50	8,800	0.27	0.70	1.22	1.98	2.57
30	0.60	7,395	0.37	0.93	1.56	2.43	3.10
25	0.71	6,223	0.51	1.20	1.95	2.95	3.70
20	0.84	5,232	0.70	1.52	2.39	3.53	4.38
18	1.00	4,400	0.94	1.89	2.89	4.19	5.14
16	1.19	3,697	1.24	2.31	3.47	4.93	5.98

*cm/s = 1.97 ft/min=14.7 gpm/ft²

Sand size and degree of fluidization is based upon the water conditions expected for the biofilter, e.g., a warmwater system will generally use larger sands than cool water systems. Smaller sands will achieve higher removal efficiencies per pass and will approach 100% ammonia removal, but often require biosolids management at the top of the sand bed column. Larger sands require much higher upflow velocities and have very short hydraulic retention times in the biofilter reactor vessel, often as short as 2 or 3 minutes. TAN removal efficiency will be in the 10 to 30% per water pass. These type of sand bed systems will not collect biosolids and require minimal maintenance. However, these beds also constantly emit fine suspended solids.

From the continuity equation, Eq. 8.11, the chosen upflow velocity, V_{sand}, and the calculated cross sectional area of the sand bed, A_{bed}, the flow required for fluidization is defined:

$$Q_{sand} = V_{sand} \cdot A_{bed} \tag{8.11}$$

This flow rate must be compared to the other flow rate calculations that were performed to maintain fish water quality conditions, Q_{wq}, at some design or target level for the overall system, e.g. ammonia, oxygen, CO_2, or solids control. The design flow rate chosen for the tank system is the largest of all of these. If one of the Q_{wq}'s is larger than Q_{sand}, then one must determine if this larger Q will result in a sand bed expansion that is too large. If so, then the sand bed cross section must be increased. Generally, one should make the sand bed cross sectional area as large as possible, again for the reason of maximizing the sand bed volume and minimizing the nutrient loading per unit sand bed particle. This becomes an iterative approach and is often confusing to the first time designer.

Sand Specifications and Selection

Suppliers of graded filter sands usually report the effective size (D_{10}) and uniformity coefficient (UC) of their sand. Fish farmers purchase sand for fluidized-bed biofilter by specifying either an effective size or some range of sieve sizes, Table 8.12, in addition to an acceptable uniformity coefficient for the sand (where smaller UC's result in sands with less variation in particle diameter). Specifying sand that is sized through a 20/40 mesh means that the largest sand passes a 20 mesh sieve (D_{eq} = 0.841 mm) and the smallest sands are retained on a 40 mesh sieve and are larger than 0.42 mm. There is always some small percentage of sands that can be considered dust, but this should be only 1% to 3% of the total mass of sand. Upon start-up of a new fluidized-bed biofilter, the bed will flush the small sand from the system for several days. Several

culture tank flushings are required to clear the fines from the system, which should be completed before fish are added to the system.

Table 8.12 Opening Sizes of U.S. Sieve Series Designation Number (Perry and Chilton, 1973)

Sieve Designation Number†	Size of Opening, (mm)	Sieve Designation Number†	Size of Opening (mm)
4	4.76	35	0.500
5	4.00	40	0.420
6	3.36	45	0.354
7	2.83	50	0.297
8	2.38	60	0.250
10	2.00	70	0.210
12	1.68	80	0.177
14	1.41	100	0.149
16	1.19	120	0.125
18	1.00	140	0.105
20	0.841	170	0.088
25	0.707	200	0.074
30	0.595	230	0.063

† Number of meshes per inch.

The ***effective size*** (D_{10}) is defined as the opening size, which will pass only the smallest 10%, by weight, of the granular sample. The D_{10} provides an estimate of the smallest sand in the sample and is the size used to estimate the maximum expansion at a given water velocity.

The ***uniformity coefficient*** (UC) is a quantitative measure of the variation in particle size of a given media and is defined as the ratio of D_{60} to D_{10}.

The D_{90} is the sieve size for which 90% of the grains by weight are smaller. The D_{90} provides an estimate of the largest sand in the sample and is the value used during design to calculate the water velocity required to fluidize even the largest sand to some minimal expansion, e.g., 20%. One should be sure to check that the design fluidization value meets this minimum requirement. The D_{90} can be estimated from the effective size (D_{10}) and the uniformity coefficient UC:

$$D_{90} = D_{10} \cdot \left(10^{1.67 \cdot \log(UC)}\right) \tag{8.12}$$

The ***mean size*** (D_{50}) is the sieve size for which approximately 50% of the grains by weight are smaller. The D_{50} provides an estimate of the average size of the sand in the sample and is the value used during design to estimate the average bed expansion at a given superficial velocity.

Using the uniformity coefficient of the sand, you can approximate the mean sand size.

$$D_{50} = D_{10} \cdot \left(10^{0.83 \cdot \log(UC)}\right) \tag{8.13}$$

If desired, the specific surface area of the static sand bed (S_b) can be approximated from the static bed void fraction ($\varepsilon \approx 0.45$) and the *sphericity* of the sand ($\Psi \approx 0.75$):

$$S_b = \frac{6 \cdot (1-\varepsilon)}{\Psi \cdot D_{50}} \tag{8.14}$$

Biosolids Management

The disadvantage of the smaller sand sizes is flocculation growth in the fluidized sand bed, especially as water temperature increases (Thomasson, 1991; Monoghan et al. 1996; Timmons, unpublished data). Biofilm growth on sand decreases individual particle effective density, which causes these particles to migrate toward the top of the biofilter column, and increases the total biofilter expansion. Heterotrophic growth continuously occurs and tends to trap sand particles in its growth. This biological growth plus the dead bacteria from biofilms also migrates to the top of the sand column. The smaller sands can be trapped in this flocculant material and remain at the top of the sand column. When the biosolid-coated sands remain at the top of the column, the shearing of biofilm material from the sand particles that occurs at the bottom of the bed near the orifice on the horizontal laterals does not occur.

If the fluidized-bed biofilter reactor vessel has translucent walls, one can easily observe the interface between the fluidized sand and the flocculant layer. When using small sands, D_{10} <0.42 mm (sieve size #40), in warmwater systems, the growth of the flocculant layer must be actively managed, or it can become uncontrollable. The fluidized-bed biofilters operator must have a regular routine of removing the flocculant layer or the whole sand bed could become engulfed. Removing the flocculant layer will also generally require replacement of sand, especially when fine sands are used. Replacement of sand has an inherent disadvantage beyond the obvious problems (sand cost and labor) in that the fine material introduced with the new sand will foul the water column. Depending upon the fish species and the rigor of the required water quality, pre-flushing of the new sand may be required.

Cornell University research experience when using sands that have D_{10} >0.42 under warmwater conditions has been that the beds have no

appreciable collection of flocculant material at the top of the fluidized-bed biofilters. This is a large advantage in terms of simplifying management. The ***disadvantage*** is that the larger sands require higher fluidization velocities and this reduces the size of the biofilter.

> **"Rule of Thumb"**
> Well managed FSB's will lose ¼ to a full tank volume of sand each year

For nearly 20 years, the Freshwater Institute has consistently managed fine sand (D_{10}= 0.20–0.25 mm) fluidized-bed biofilters in several recirculating rainbow trout culture systems. The fluidized-bed biofilters operated reliably during this period, with TAN removal efficiencies typically ranging from 70–90%. Biosolids growth in the fluidized-bed biofilters was usually controlled by siphoning biosolids from the top of fluidized-bed biofilters as their beds reach a maximum depth, and then replacing lost sand as needed. The Freshwater Institute has also controlled biofilm thickness by shearing the biofilm in-vessel, using a pump to transport the flocculant particles from the top of the biofilter to the bottom of the bed, where shear forces are greatest. This approach to controlling biofilm growth effectively maintained bed expansion at a fixed level without significant flushing of sand. All fluidized bed sand filters no matter how well managed will lose approximately ¼ to a full tank volume of sand per year. These sand losses may be insignificant on a daily basis, but over the course of several months to one year will generally require the replenishment of sand. Fortunately, sand is inexpensive. In particular, circumstances, pro-active capture of sand particles for recycling back into the vessel may be warranted.

In the Freshwater Institute's coldwater recycle system, it was found that biosolids did not accumulate within expanded beds using sands with effective sizes of (D_{10}) 0.60 and 0.80 mm. However, biosolids did sometimes collect in a distinct layer above the expanded sand layer. Siphoning the biosolids layer was simple with these larger sands, because the expansion depth of these sands remained fairly constant and the biosolids could be removed relatively free of sand. Because the biosolids layer is expanded, it is also fluid, which greatly reduces sand loss and the need to replace old sand with new sand (when larger sands are used). A siphon can be strategically placed at a height so that only biosolids are removed and not clean nitrifier-biofilm-covered sand. A distinction is being made here between biosolids that are sand particles

encased within bioflock and heterotrophic bacteria and sand that is covered with biofilm from nitrifiers.

Design the Water Delivery System

Designing an effective fluidized-bed biofilter water delivery system requires appropriate sizing of the plenums, pipes, and orifices to promote equal fluidization of the sand bed. Generally, this involves using a collector plenum to receive water from the pumps and then routing a given number of laterals from this plenum to provide an effective lateral grid across the bed floor. Maximum spacing of laterals (center to center) is in the 15 to 30 cm (6 to 12 inch) range. The basic idea is that the cross sectional area of the pipe upstream of a group of laterals or laterals upstream of orifices should be 2 times the collective area of the downstream flow area of the laterals or downstream area of the orifices. This results in the pressure gradient being dominated by the loss across the reduction section so that each reduced section, e.g., lateral or orifice, will see approximately the same pressure regardless of its location along the lateral (orifice case) or lateral attached to a collection plenum/manifold. This approach ensures that the flow will be approximately the same to each of the laterals from the manifold/plenum or from a lateral to each orifice.

The following area ratios should be maintained between manifolds and laterals and between laterals and orifices:

$$1.5 < \frac{\text{manifold area}}{\text{lateral area}} < 3 \tag{8.15}$$

$$2.0 < \frac{\text{lateral area}}{\text{orifice area}} < 4 \tag{8.16}$$

As a rule of thumb, the flow distribution manifolds designed according to these criteria have a total pressure requirement (in meter or feet of water pressure) equal to approximately three times the sand depth plus the height that the water must be lifted from the floor sump to the top of the biofilter. Therefore, total pumping head requirements range from 5 to 11 m (16 to 36 ft), depending mostly on the depth of the biofilter column. For precise pressure estimates, the hydraulic grade line should be calculated across the piping from the pump sump to the water depth in the FSB column.

Total head loss through the FSB treatment device is the sum of dynamic losses through the manifold piping, losses from fluidizing the sand bed (estimated as 0.9 times the static depth of the sand) and the elevation difference between the water inlet and free surface at the top of the FSB. Summerfelt (1996), Summerfelt (2006) and Summerfelt et al. (1996) give a more detailed discussion on how to estimate pressure losses through the FSB system and how to size laterals and plenums/manifolds. An example of how a spreadsheet can be used to size the pipe manifold, pipe laterals, and distribution orifices is provided using the equations presented in this chapter can be found in Table 8.13.

Sizing of Orifices

Orifice size is selected to fluidize the bed by assuming the pressure loss across the orifice is greater or equal to the static sand depth:

$$\frac{C_{orifice} V^2_{orifice}}{2g} > h_{sand} \tag{8.17}$$

Orifice coefficient values, $C_{orifice}$, are in the range of 0.6 to 0.8. While the choice of this orifice coefficient does not appear to be terribly significant, since the known value in the equation will be the flow rate, small changes in $C_{orifice}$ can significantly affect the estimate of the pressure loss term. The severe negative consequence of using a $C_{orifice}$ value that is too low is that the bed may not fluidize in all regions. A $C_{orifice}$ value of 0.7 has been used in past designs, or a $C_{orifice}$ value of 0.6 and an orifice size is selected to create a pressure loss 1.2–1.5 fold greater than the sand depth.

Plugging Concerns

A major concern in operating fluidized-bed biofilters is the plugging of the lateral systems with sand. This can happen if the check valves in line with the pumps and biofilter malfunction and do not close at the time pumps are shut down. Check valve failure should be rare, but if it ever does happen, water will siphon out of the biofilter to the pump sump (which is at a lower elevation) and carry sand into the pipe laterals until the pipes are plugged. To prevent this, premium quality swing check valves should be used to reduce the chance of failure. In the event of a failure, laterals can be unplugged, usually in a matter of hours, with the use of clean-outs on the pipe laterals at the top of the biofilter. One must anticipate that the pipe laterals will at some point become plugged, and

know how to clean them effectively if the biofilter is to be restored to operating mode.

Table 8.13 Spreadsheet Example of Fluidized Sand Bed (FSB). Design process spreadsheet is used to calculate the size of the FSB manifold, pipe laterals, and distribution orifices. Values in **BOLD** text are user assigned numbers, entered in an iterative procedure until the criteria in ***BOLD Italics*** are met.

Design Parameter		Comments
Expansion velocity, cm/s	**2.6**	
Expansion velocity, ft/min	5.1	
Flow Rate, gpm/ft^2	38.0	
Diameter of sand tank, inches	72	
Diameter of sand tank, ft	6.0	
Area of floor cross section, ft^2	28.3	
Total flow, gpm	1,074	
Lateral pipe diam, inches	**3.00**	
Lateral pipe area, sq inches	7.07	
Lateral Length, avg, feet	4.80	note: for round tank, avg L is 86% of vessel Diameter
Lateral spacing, inches	**12**	
Number of laterals (calc)	7.0	
Number of Laterals assigned	**6**	you must have a unit number
Orifice diameter, inches	**0.563**	
Orifice area, in^2	0.248	
Orifice spacing (='s lat space) inches	12	
Number of orifices/lateral	11.6	assigned in pairs
Total # of orifices assigned per lateral	**12**	
Orifice area per lateral, in^2	2.99	
Total Orifice area, ft^2	0.124	
Pipe area: orifice area	2.4	recommended between 2 to 4
Manifold area: lateral pipe area	**1.5**	recommended area ratio is 1.5 to 3.0
Min Req Manifold diameter, inch	9.0	using one header pipe for all the laterals
Min Req Manifold diameter, inch	6.4	using two header pipes for all the laterals
Orifice Coefficient (0.6 to 1.0)	0.8	
Velocity across orifice, ft/s	24.0	
Head loss across the orifices, ft	9.0	This number needs to be > sand depth

Currently, this siphoning action has been successfully prevented by placing swing check valves above or below the pump(s). Heavy, brass or PVC swing check valves with a solid rubber swing flap or a brass swing flap and a well-machined seat ledge were used. These valves are still working effectively after several years of operation. Others have used siphon breaks to let air into the manifold above the biofilter when pumps are shut down, which allows the water in the pipe manifold to drain into the biofilter and prevents siphoning. The Freshwater Institute has tried a siphon break such as this and found it to be undesirable, because the air filling the laterals and manifold is forced through the sand bed when pumping is restored. The large slug of air passing through the biofilter has caused significant volumes of sand and biosolids to dump out of the biofilter. This phenomenon is probably much more of a problem with the finer sands and the associated mass of biosolids that accumulate in these biofilters. Similarly, any air leak into the pumping and lateral system will also promote sand loss from a bed and prevent proper operation, especially when smaller sands are used.

When using check valves, some allowance should be made to prevent air locks in the system. Simply examine the system, and if there is the potential for air lock to occur, make provisions to allow air to escape from the pump. This is a typical problem if the pumps are submerged and the collection sump is drained once a pump is turned off. When the sump area is refilled, the pipe above the pump will have trapped air and will prevent the pump from moving enough water to open the check valve.

COST BASIS OF SAND FILTERS

As stated in the beginning of this chapter, the main argument for using fluidized-bed biofilters is that their cost per unit of TAN treated is low compared to competing technologies. This is true only for larger systems. Fingerlakes Aquaculture, LLC (Groton, NY) operated 1.83 m diameter by 4.6 m high fluidized-bed biofilter systems using a static depth of sand of 2.14 m. Volume of sand in these systems was 5.61 m^3. Using a TAN nitrification rate of 1 kg TAN/day/m^3 indicates a nitrification rate of 5.61 kg TAN per day per fluidized-bed biofilter, which would support a fish feeding load of 187 kg feed per day. More recently, Fingerlakes installed and operated 1.83 m diameter by 3.5 m high Cyclo-biofilters with good success. Fingerlakes Aquaculture uses a design value of 100 kg and 275 kg feed per day for these biofilter systems, respectively.

Costs provided by Fingerlakes Aquaculture for their 1.83 m diameter fluidized-bed biofilter tank systems are approximately $2,200 for the fiberglass reactor vessel, $800 for a fiberglass plenum/manifold, plus the costs of pipe and valves. Fingerlakes Aquaculture estimates plumbing labor requirements to be approximately 50 man-hours to fully plumb a system to the fish tank unit.

Summerfelt and Wade (1997) report that two fluidized-bed biofilters built at two commercial fish farms sized to treat flows of 1.5 to 2.3 m^3/min cost around $6,000, including biofilter vessel, sand, piping, valves, shipping, and labor for installation. These prices were approximately five times less than the estimated cost for a installing a trickling filter of similar TAN removal capacity.

SUMMARY OF STEPS

In summary, the following design steps should be followed when designing a fluidized sand bed:

1. Determine the TAN load
2. Determine the sand volume required to match TAN load
3. Select the design depth of sand bed
4. Determine the cross-sectional area of the sand bed
5. Select the sand size in relation to flow rate available or desired or determine the required flow rate to fluidize the bed of the sand available
6. Compare to the other required flow rates for maintaining water quality parameters in the fish tank; make adjustments in the sand bed cross section if one of the Q_{wq}'s is larger than Q_{sand}
7. Design the water delivery system

DESIGN EXAMPLE – FLUIDIZED-BED SAND BIOFILTER

Design a FSB for a warmwater system using 20/40 sized sand for a feeding rate of 27.5 kg feed/day.

Step 1: Determine the TAN load

$$TAN\ Load = 27.5 \frac{kg_{feed}}{day} * 0.032 \frac{kg_{TAN}}{kg_{feed}} * = 0.89 \frac{kg_{TAN}}{day}$$

Step 2: Determine the sand volume required to match TAN load

$$Volume = \frac{\dfrac{0.89\ kg_{TAN}}{day}}{\dfrac{1\ kg_{TAN}}{day \cdot m^3_{sand}}} = 0.89\ m^3_{sand}\ required$$

Step 3: Select the design depth of sand bed

$$h_{sand} = 2\ m$$

This is the unexpanded sand bed height; placed on floor level. Selection based upon ceiling height and elevation required to permit gravity flow back to culture tank through aeration column and/or oxygenation column or other equipment devices.

Step 4: Cross sectional area of bed

$$A_{bed} = \frac{V_{media}}{h_{sand}} = \frac{0.89\ m^3}{2m} = 0.45\ m^2$$

Step 5: Fluidization velocity

For a 20/40 sand, Table 8.12 can be used to estimate D_{10} (40 mesh) = 0.42 mm. If the sand has a UC of 1.5, Eqs. 8.13 and 8.14 can be used to estimate D_{90} and D_{50}:

$$D_{90} = 0.42 \cdot 10^{1.67 \times \log(1.5)} = 0.83mm$$

$$D_{50} = 0.42 \cdot 10^{0.83 \times \log(1.5)} = 0.59mm$$

Using Table 8.12, the required velocity to expand the above 20/40 sand by 50% with a D_{eq} = 0.59 mm is 1.56 cm/s (Table 8.12 has a D_{eq} of 0.60 mm, so we use this value; or you could interpolate if more accuracy is needed for some particular application). To check if the largest sands will expand, the velocity required to expand a 0.83 mm sand 20% is 1.52 cm/s (again the table has a 0.84 mm D_{eq} value, so this is what we used for this example). Since the 1.52 cm/s is less than the bed design velocity used for the D_{50} sand at 50% expansion, the velocity selection is considered safe. If the D_{90} velocity criteria resulted in a velocity that exceeded the D_{50} value, then you should increase the overall velocity and go with a larger expansion than 50%. The design example will use 2.0 cm/s as a conservative safe estimate to ensure the large sands in the

20/40 sand used will fluidize. And going back to Table 8.12 and for the D_{50} value of 0.59 mm, the bed expansion is predicted as being around 75%. This expansion is considered a very safe value to ensure full mixing and is therefore consistent with our recommended conservative approach.

Step 6: Required Flow Rate

Given the selection of an expansion velocity of 2.0 cm/s and the cross sectional area of the bed (0.45 m²), the required flow rate is:

$$Q_{sand} = V_{sand} * A_{bed} = 0.020\frac{m}{s} * 0.45\,m^2 * \frac{60\,s}{\text{min}} * \frac{1000\ l}{1\ m^3} = 540\ lpm$$

Again, here is a good example where the flow requirements for a particular criteria (in this case, fluidization of the sand) is much less than the controlling critera from some other criterion, which in the current common design example is 1514 Lpm for tank exchange rates. If we employed that flow rate on our sand filters, we would blow the sand out of the reactors. So, in this case, only some of the required flow (540 Lpm) would be pumped through the sand filter and the rest of the flow would bypass the biofilter. Other biofilters can accept higher flow rates without a problem, e.g., trickling filter, moving bed biofilters, or microbed filters.

In a coldwater application, sands with D_{10} of around 0.25 mm are selected because of the high nitrification removal efficiency across the filter that can be achieved, due to the high hydraulic retention time of the vessel. These biofilters are usually oversized because the sand selection locks in a velocity of near 1.0 cm/s and the recirculating flowrate is controlled by the oxygen requirements of the fish. In this case, you solve for A_{bed} knowing Q_{wq} and V_{sand}:

$$A_{bed} = \frac{Q_{wq}}{V_{sand}}$$

A design of a fluidized sand filter to meet the design objectives of this example (design feeding load of 27.5 kg/day or 60 lb/day) is given in Table 8.13.

8.8 DESIGN EXAMPLE: MICROBEAD BIOFILTER

Design information for dimensioning or sizing a microbead biofilter is presented by Greiner and Timmons, (1998) and Timmons, et al. (2006). In practice, TAN removal efficiency is often empirically determined for a fixed set of successful conditions such as fish species, feed load, specific filter design, and TAN influent concentration. The simplest case is when the TAN removal efficiency for commercial scale microbead filters is known, based on actual performance data. The required total nitrification surface area (A_{media}, m^2) is then calculated from the microbead filter TAN load (P_{TAN}, kg/day) and the estimated nitrification rate (r_{TAN}, g TAN/m^2/day). The bioreactor volume (V_{media}, m^3) is a function of the total filter surface area (A_{media}, m^2) and the specific surface area (SSA, m^2/m^3 biofilter media) of the filter media.

The design of a microbead follows the same set of steps as the trickling tower with the nitrification rate based on the active surface area of the media. We will design a microbead filter by continuing the design process for a single Omega fish facility fingerling/growout Pod utilizing a microbead biofilter whose media (Type B) has a SSA of 2520 m^2/m^3 (770 ft^2/ft^3). The water flow requirements are currently based on the tank water exchange rates that require 90.8 m^3/hr (1514 Lpm or 400 gpm).. The sizing of the microbead biofilter is based on the daily TAN production rate, which is directly dependent on the feeding rate of 27.5 kg/day. Microbead filters also require a minimum hydraulic loading rate 1290 m^3/m^2 d (22 gpm per ft^2) cross sectional area. This often exceeds other flow design criteria.

The design of a microbead filter follows the same set of steps as the trickling tower with the nitrification rate based on the active surface area of the media. Using the initial design parameters of the trickling tower design example, the process is the same up to Step 4.

Step 4: Calculate volume of media (V_{media}) required to remove P_{TAN} from the Volumetric TAN removal rate (VTR). Based on experience with the commercial systems, the estimated Volumetric TAN removal rate is 1.2 kg TAN/m^3 day (Timmons, et al., 2006).

$$V_{media} = \frac{P_{TAN}}{VTR}$$

$$= \frac{0.885\ kg\ TAN}{1.2\dfrac{kg\ TAN}{m^3}} = 0.74\ m^3$$

Step 5: Calculate the biofilter cross-sectional area ($A_{biofilter}$), assuming a maximum depth of the filter, D_{media}, of 0.45 m.

$$A_{biofilter} = \frac{V_{media}}{D_{media}}$$

$$= \frac{0.74\ m^3}{0.45\ m} = 1.64\ m^2$$

Check that the hydraulic loading rate; HLR of 1290 m^3/m^2 day (22 gpm/ft^2) required to prevent clogging of the media bed meets the required flow for the fish oxygen demand (Q_{tank}).

$$Q_{tank} = 1290\frac{m^3}{m^2 day} * 1.64\ m^2$$

$$= 2{,}120\frac{m^3}{day} = 1{,}470\ Lpm = 390\ gpm$$

Finally recheck the tank exchange rate. (volume of two growout tanks and two fingerling tanks is 57.2 m^3, see Table 5.14):

$$Tank\ exchange\ rate = \frac{V_{tank}}{Q_{tank}} = \frac{57.2\ m^3 \cdot \dfrac{1{,}000 L}{m^3}}{1470\dfrac{L}{\min}} = 39\ \min$$

The tank exchange rate (39 minutes) compares reasonably well with our current flow requirement for tank water exchange established when we did our first biofilter design example, for the Trickling Filter design. Depending upon the scale and loading of the system, in some cases the biofilter may require more flow that is required for tank exchange in the fish tank. Under these cases, either you can operate the tank at these higher flow rates or some of the flow (excess) can be recycled from the biofilter to the inlet side, providing both the required hydraulic loading rate and reducing the total flow through the production tank.

Step 6: Calculate the biofilter diameter ($D_{biofilter}$) from the biofilter cross-sectional area ($A_{biofilter}$) and volume (V_{media}).

$$D_{biofilter} = \sqrt{\frac{4 \cdot A_{biofilter}}{\pi}} = \sqrt{\frac{4 \cdot 1.64\ m^2}{3.14}} = 1.45\ m = 4.75\ ft$$

It is critically important with microbead filters to remove as much of the suspended solids to prevent clogging of the media. To accomplish this, rotating drum filters with a mesh size of 60 to 200 μm (micron) are often used ahead ofthe microbead filter.

8.9 Design Example: Moving Bed BioReactor

For sizing a Moving Bed BioReactor (MBBR) in aquaculture, only limited information is available (Rusten et al., 2006). In practice, TAN removal efficiency is often empirically determined for a fixed set of successful conditions such as fish species, feed load, filter height, filter media type, hydraulic surface load, suspended solids unit, and TAN influent concentration.

The simplest case is when the TAN removal efficiency for a certain MBBR filter influent concentration is known, based on data for a fixed filter volume, media type, hydraulic loading, TAN removal rate, and temperature. The required total nitrification surface area (A_{media}, m^2) is calculated from the MBBR filter TAN load (P_{TAN}, kg/day) and the estimated nitrification rate (r_{TAN}, g TAN/m^2/day). The bioreactor volume (V_{media}, m^3) is a function of the total filter surface area (A_{media}, m^2) and the specific surface area (SSA, m^2/m^3 biofilter media) of the filter media. The shape of the reactor depends on the Height to Diameter ratio defined for the reactor vessel.

The design of a MBBR follows the same set of steps as the trickling tower with the nitrification rate based on the active surface area of the media. Using the initial design parameters of the trickling tower design example, the process is the same up to Step 4 (TAN design load of 0.89 kg TAN/day). If your design maximum loading rate for MBBR is above 50 kg/day, then these applications will most probablyrequire more sophisticated mixing systems beyond simple aeration. In these cases, you should probably seck expert assistance from one of the media manufacturer companies or other capable consulting companies with specific experience and success in design these larger scale operations.

Step 4: Calculate volume of media, V_{media} based on the volumetric Nitrification Rate (VTR) associated with the media being used, for

example, Curler Advance X-1 has a VTR of 605 g TAN/m^3day (17.14 g TAN/ft^3day) for growout systems at 25 to 30°C.

$$V_{media} = \frac{P_{TAN}}{VTR}$$

$$= \frac{0.885\ kg\ TAN\ /\ day}{0.605\ \frac{kg\ TAN}{m^3 \cdot day}} = 1.46\ m^3$$

Note that for coldwater applications (12–15°C) when TAN concentrations entering the MBBR are approximately 1–2 mg/L, then the Volumetric TAN removal rate is reduced to 468 g TAN/m^3 day (13.26 g TAN/ft^3).

Step 5: Calculate the biofilter cross-sectional area given a height/diameter ratio of 1.0. This ratio can vary between 1.0-1.2 and depends mostly on the effectiveness of mixing and aeration. The diameter should not exceed 2 m (6.5 ft). In this example, a 60% fill rate was chosen that yields a reactor tank volume of 5.0 m^3. If additional nitrification is required, additional media could be added up to 70% fill.

$$D_{biofilter} = \sqrt[3]{\frac{4 * V_{biofilter}}{\pi * 1.0}} = \sqrt[3]{\frac{4 * 1.46 m^3}{3.14 * 1.0}} = 1.37\,m$$

Given a height/diameter ratio of 1.0, yields a reactor height of 1.37 m (4.5 ft). Thus a 1.5 m (5 ft) diameter tank, 1.5 m (5 ft) tall, with the addition of some free board, would be required. There are several manufacturers of closed lid polyethylene tanks that would work well for this application. Inlet is a simple pipe discharge near the bottom of the tank and the outlet can be either a slotted pipe or a screen discharged to prevent media from escaping. A closed cover is required for large biofilters to allow off gassing of carbon dioxide production outside of the building. Aeration and mixing requirements would be approximately 5 times the reactor volume in m^3/hr with the addition of a 50% spare capacity.

$$\dot{V}_{air} = \frac{5}{hour} * V_{biofilter} = 5/_{hour} * 5m^3 = 25m^3/_{hour}$$

$$25m^3/_{hour} * 24\ hour/_{day} * \left(1.81\ kgTAN/_{day}\right)^{-1} = 331.5m^3\ air/_{kgTAN}$$

Aeration Requirements. In this example we have a total air requirements of 1.12 cf/g TAN removed. It is important to note here that the amount of media required, tank volume and airflow rate are all directly related to the rate of TAN removal required. This could be provided with either coarse bubble diffusers or pipe diffusers with 3 mm holes. A chart is given (see also Figure A.22, pg 919) to provide guidelines on required pipe diameters for specific airflow requirements. These are guidelines only.

Apart from nitrification, a MBBR should provide for some removal of carbon dioxide. It is important with MBBR to remove as much of the suspended solids to prevent clogging of the media and a means for settling out accumulated solids are provided.

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List of Symbols

A	Area, m^2
$A_{biofilter}$	Biofilter cross-sectional area, m^2
A_{media}	Surface area required to remove P_{TAN}, m^2
a_{TAN}	TAN production per unit of feed fed, kg TAN/kg feed
a_{DO}	DO demand per kg of feed fed, kg O_2/kg feed
ATR	Areal TAN conversion rate, mg TAN/m^2 day
$C_{orifice}$	Orifice coefficient
D_{10}	Sand particle effective size, smallest 10% by weight of sample
D_{50}	Sand size for which 50% of the grains by weight is smaller, and also called mean size of the sand
D_{90}	Sand sizes for which 90% of the grains by weight are smaller
$D_{biofilter}$	Diameter of a biofilter tank, m

D_{eq}	Equivalent diameter of a sand particle assuming a spherical shape for the sand particle of equal mass
$Depth_{media}$	Depth of media in biofilter, m
DO_{inlet}	DO concentration at inlet to fish tank, mg/L
DO_{tank}	Dissolved oxygen concentration in the fish tank, mg/L
DO	Dissolved oxygen concentration, mg/L
f_{rem}	TAN removal efficiency across the biofilter as a fraction
g	Acceleration of gravity, m/s^2
h_{sand}	Height of unexpanded sand column, m
HLR	Hydraulic loading rate, gpm/ft^2 ($m^3/m^2 \cdot$ day)
HRT	Hydraulic retention time, minute
I_s	*In situ* nitrification fraction, unitless
M_{fish}	Biomass of fish in culture tank, kg
P_{TAN}	TAN production by fish, kg/day
Q	Flow rate, m^3/s
Q_{sand}	Flow rate required to fluidize the sand bed, m^3/s
Q_{tank}	Water flow requirement for fish DO demand, m^3/s
Q_{wq}	Flow rates required to maintain water quality design values in the fish rearing tank, m^3/s
r_{feed}	Feeding rate to fish on a % per day of bodyweight,%/day
r_{TAN}	Nitrification rate, g TAN/m^2/day
R	Relative ammonia removal rate per unit time
R_{feed}	Feeding rates of the peak feeding, (kg/day)
R_{DO}	Dissolved oxygen requirement, kg/day
R_{flow}	Fraction of water that is reused
R_{TAN}	Ammonia nitrogen production rate, g TAN/day
RBC	Rotating biological contactor (acronym)
S_b	Specific surface area per unit particle volume, cm^{-1}
SSA	Specific surface area, ft^2/ft^3 (m^2/m^3)
T	Water temperature, °C
TAN	Total ammonia nitrogen
TAN_{out}	Concentration of TAN leaving fish culture tank, mg/L
UC	Uniformity coefficient (related to sand characteristics)
V_{sand}	Upflow velocity through sand bed, m/s or cm/s
v_f	Floating bead filter sizing criterion, kg feed/m^3 of media·day
V_{media}	Volume of media required to remove P_{TAN}, m^3
$V_{orifice}$	Velocity at orifice hole in a lateral, m/s
V_{tank}	Volume of tank, m^3 (gal)
$Volume_{beads}$	Volume of bead media required for a specific application, m^3
VTR	Volumetric nitrification rate, g TAN/$m^3 \cdot$ day

ε Static bed void fraction
ρ Fish biomass density in culture tank, kg/m^3 (lbs/gal)
Ψ Sphericity of the sand

CHAPTER 9

DENITRIFICATION[1]

9.0 INTRODUCTION

Historically, nitrate (NO_3), the end product of nitrification, has not been of major concern in RAS due to its low toxicity to freshwater organisms (Lee et al., 2000). However, with the high degree of water reuse that is inherent in both freshwater and especially marine recirculating aquaculture systems, NO_3 reduction becomes more important, since accumulations as high as 100-1,000 mg NO_3-N are not uncommon. Tucker (1999) suggested nitrate nitrogen levels be maintained lower than 20 mg/L for marine larval and juvenile fish and 50 mg/L for older fish. Reported maximum nitrate concentrations in recirculating systems are as high as 400-500 mg NO_3-N/liter (Otte and Rosenthal, 1979; Honda *et al.*, 1993). Hrubec et al. (1996) reported on work with hybrid striped bass that supported the theory that prolonged exposure to elevated levels of nitrate may decrease the immune response, induce hematological and biochemical changes indicative of a pathologic response, and may increase mortality. High nitrate concentrations can adversely affect the growth of commercially cultured aquatic organisms, among including: eel (Kamstra and van der Heul, 1998), octopus (Hyrayama, 1966), trout (Berka *et al.*, 1981) and shrimp (Muir *et al.*, 1991). Of great significance is the toxicity of nitrate to marine organisms due to its inhibitory effect of the animal's osmoregulatory ability (Russo and Thurston, 1991). The osmoregulatory stress can manifest itself in the form of inhibition of reproductive cycles, poor egg development, delayed hatching times and higher mortalities and/or inhibition of growth rates of animals of all ages (Shimura et al., 2002). In marine species, the concentration at which nitrate is toxic, lethal or sub-lethal effects that

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have been detected can vary from 2.2 to more than 5,000 mg/L-N (NGSOWP-CDECO, 2003).

Nitrate is very stable in the natural environment and can be a source of pollution on the receiving waters, causing eutrophication and algal blooms and the resultant high dissolved oxygen consumption as the algae die off. In the United States, the EPA has limited the nitrate and nitrite concentration in potable water to 10 mg/L-N and 1 mg/L-N respectively and has placed its control on the priority list. While denitrification may be expensive, it may be mandatory for many freshwater and most saltwater facilities with either limited water supply or stringent discharge requirements. In the present chapter, the fundamentals of denitrification as well as the application of this process in recirculating aquaculture systems are discussed.

9.1 BACKGROUND

Biological denitrification is the microbial reduction of nitrate (NO_3^-) or nitrite (NO_2^-) to nitrogen gas (N_2) by heterotrophic and autotrophic facultative aerobic bacteria and some fungi widely found in the environment. A relative broad range of bacteria can use either nitrate or oxygen to oxidize organic material, easily shifting between oxygen respiration and nitrogen respiration, depending upon the oxygen concentration. Denitrifiers are common among the heterotrophic, Gram-negative *Proteobacteria*, such as *Pseudomonas*, *Alcaligenes*, *Paracoccus*, and *Thiobacillus*. Some Gram-positive bacteria, including *Bacillus*, can denitrify (Rittman and McCarty, 2001). In addition to heterotrophic microorganisms, several autotrophic microorganisms are also capable of denitrification. These organisms, which use reduced inorganic sulfur and iron compounds or hydrogen as their electron donor and derive carbon from an inorganic source, are often dominant in organic-poor environments where such donors are present.

Under anoxic conditions, nitrate and nitrite replace oxygen as the electron acceptor for the oxidation of a wide variety of organic or inorganic electron donors. The term anoxic defines the conditions optimal for denitrification, low oxygen and high nitrate.

Denitrification proceeds in a stepwise manner in which nitrate (NO_3^-) is sequentially reduced to nitrite (NO_2^-), nitric oxide (NO), nitrous oxide (N_2O) and finally N_2 gas.

$$NO_3^- + 2e^- + 2\,H^+ \rightarrow NO_2^- + H_2O \tag{9.1}$$

$$NO_2^- + e^- + 2\,H^+ \rightarrow NO + H_2O \tag{9.2}$$

$$2\,NO + 2e^- + 2\,H^+ \rightarrow N_2O + H_2O \tag{9.3}$$

$$N_2O + 2e^- + 2\,H^+ \rightarrow N_2\,(g) + H_2O \tag{9.4}$$

The half-reaction equation for denitrification (not taking into account the carbon requirements for bacterial growth and maintenance) for nitrate and nitrite as electron acceptors are as follows (EPA, 1993):

$$\tfrac{1}{5}NO_3^- + \tfrac{6}{5}H^+ + e^- \rightarrow \tfrac{1}{10}N_2 + \tfrac{3}{5}H_2O \tag{9.5}$$

$$\tfrac{1}{3}NO_2^- + \tfrac{4}{3}H^+ + e^- \rightarrow \tfrac{1}{6}N_2 + \tfrac{2}{3}H_2O \tag{9.6}$$

Reduced organic carbon which is oxidized during heterotrophic denitrification can be generally presented as:

$$\tfrac{1}{4}\,CH_2O + \tfrac{1}{4}\,H_2O \rightarrow \tfrac{1}{4}\,CO_2 + H^+ + e^- \tag{9.7}$$

Overall transformation yields:

$$NO_3^- + 6\,H^+ + 5e^- \rightarrow \tfrac{1}{2}\,N_2 + 3\,H_2O \tag{9.8}$$

This process can be optimally mediated by denitrifying bacteria under specific conditions, such as: (1) low redox potential, (2) low oxygen levels, (3) availability of an organic carbon source used as an electron and carbon donor, (4) a nitrate source as an electron acceptor, (5) an optimal pH range of 7.0-8.5 and (6) an optimal temperature range of 25-32 °C (Park, 2000). If the system is not controlled properly however, low redox conditions can develop, which encourage the production of toxic hydrogen sulfide (Otte and Rosenthal, 1979). Lower limits for nitrate-nitrogen concentrations range from 10 to 50 mg/L before hydrogen sulfide can become a problem.

Heterotrophic Denitrification

There has been a great deal of research conducted on denitrification in the wastewater treatment industry and to a limited extent in intensive recirculating aquaculture systems. Although there are numerous possible sources for the organic electron donor, only a few simple organic

substrates have been extensively studied and used in the wastewater treatment industry. These were utilized as exogenous electron donors and carbon sources because there were insufficient quantities in the wastewater being treated, such as in advanced treatment of secondary effluents or potentially aquaculture systems. Simple, commonly used compounds that could be purchased in bulk at low prices were evaluated such as methanol, acetate, glucose, and ethanol. Methanol (CH_3OH) has seen widespread use, primarily because it was cheap and readily available. The disadvantages of using methanol are the safety issues associated with its transportation, handling and storage as it is considered a reactive and toxic compound. In aquaculture, several potential electron donors have been researched and evaluated for their performance. For example, a recently reported project at the Mote Marine Lab, Center for Aquaculture Research and Development (Hamlin et al., 2008) evaluated four carbon sources on the performance of commercial scale denitrification reactors: methanol, acetic acid, refinery molasses, and starch.

The stoichiometric equation for methanol as the exogenous electron donor is as follows:

$$NO_3^- + 1.08\ CH_3OH + 0.24\ H_2CO_3 \rightarrow 0.056\ C_5H_7NO_2 + 0.467\ N_2 + 1.68\ H_2O + HCO_3^- \quad (9.9)$$

Equation 9.9 suggests that 2.47 g of methanol are required to reduce 1 g of nitrate-nitrogen. Experimental ratios of methanol to nitrate range from 2.5 to 3 g methanol/g nitrate nitrogen. In addition, for each 1 g of nitrate nitrogen removed, 0.45 g of VSS is generated, plus 3.57 g of alkalinity as $CaCO_3$ or one equivalent of alkalinity per g of nitrate-N denitrified.

Sophisticated and costly computer control systems are often required to regulate the carbon dosage to prevent overdosing of carbon (Lee et al., 2000; Boley et al., 2000). An excess of carbon in the absence of nitrate in an anaerobic environment can reduce the redox potentials promoting the reduction of sulfates and the production of toxic sulfides (Whitson et al., 1993). These methods also require multiple treatment components, further increasing the overall cost. Finally, there are certain risks associated with conventional denitrification technologies, such as failure of the process control system, which can lead to overdosing of the carbon source and/or the production of hydrogen sulfide (Whitson et al., 1993).

In addition to methanol, several other carbon sources have also been evaluated in commercial scale denitrification reactors in a large scale sturgeon growout operation (Hamlin et al., 2008). These include acetic

acid, refinery molasses, and starch. The denitrification bioreactors consisted of a 1.89 m^3 covered conical bottom polyethylene tank containing 1.0 m^3 media through which water up-flowed at a rate of 10 Lpm. Nitrate-nitrogen concentrations were effectively reduced to zero at influent concentrations ranging from 11 to 57 mg/L NO_3-N. In addition, to the stoichiometry for denitrification, the stoichiometry for dissolved oxygen removal was also reported (Hamlin et al., 2008), since in some cases the removal of oxygen is accomplished by the carbon source.

The stoichiometric equations for denitrification and oxygen reduction with methanol are shown below: (McCarty et al, 1969):

$$O_2 + 0.93\ CH_3OH + 0.056\ NO_3^- + 0.056\ H^+ \rightarrow 0.056\ C_5H_7NO_2 + 0.65\ CO_2 + 1.69\ H_2O \quad (9.10)$$

$$NO_3^- + 1.08\ CH_3OH + H^+ \rightarrow 0.065\ C_5H_7NO_2 + 0.47\ N_2 + 0.76\ CO_2 + 2.44\ H_2O \quad (9.11)$$

Finally, the digestion of organic sludge and manures in recirculating systems forms an excellent carbon source for denitrifiers with the following stoichiometric relationship:

$$C_7H_{13.4}O_{3.5}P_{0.3}N + 3.3NO_3^- + 2.6H^+ \rightarrow 1.6N_2 + 2.9CO_2 + 0.2NH_3 + 4.9H_2O + 0.8C_5H_7O_2P_{0.1}N + 0.2\ PO_4^{3-} \quad (9.12)$$

Autotrophic Denitrification

Inorganic electron donors can also be used and are gaining in popularity, especially in public marine aquariums. Hydrogen gas is an excellent electron donor with a relative lower cost compared to other organic compounds that produce more biomass as a result of heterotrophic growth. The main disadvantage of hydrogen has been the lack of a safe and efficient transfer system. Recent developments in membrane-dissolution devices overcome the explosion hazard and makes hydrogen gas a viable alternative (Rittmann & McCarty, 2001). The stoichiometric relationship for using hydrogen gas to remove nitrate-nitrogen is:

$$2NO_3^- + 5H_2 \rightarrow N_2 + 4H_2O + 2OH^- \quad (9.13)$$

Popular in commercial and public aquariums is the use of reduced sulfur in its elemental form to drive autotrophic denitrification. Sulfur is oxidized to SO_4^{2-} and supplied in a solid matrix or pellet that includes a

solid base, such as $CaCO_3$. The base is added because during the oxidation process a strong acid is generated and the base prevents shifts in the pH. The stoichiometry for nitrate nitrogen removal using elemental sulfur is:

$$10NO_3^- + 11S + 4.1HCO_3^- + 0.5CO_2 + 1.71NH_4^+ + 2.54H_2O \rightarrow 0.92C_5H_7NO_2 + 11SO_4^{2-} + 5.4N_2 + 9.62H^+ \quad (9.14)$$

9.2 Unit Processes for Denitrification

Many of the treatment processes employed in aquaculture are adaptations of unit processes used in the wastewater treatment industry. Over the years, these have been modified to conform to the higher water quality standards in aquaculture, lower concentration of ammonia-nitrogen and BOD, and to make them as cost effective as possible. These processes for aquaculture applications include for example the use of sedimentation basins and microscreen filters for solids capture; trickling filters, RBCs and fluidized bed biofilters for nitrification. Thus, it seems appropriate to examine denitrification technologies employed by current state-of-the-art wastewater treatment facilities. Because of the recent requirements in the USA for nitrogen removal from the discharge of wastewater treatment plants, several options have been developed over the past 10 years based on the suspended sludge, denitrification process and more recently fixed film bioreactors. These have been included in the sequence of unit processes used to treat the wastewater either by using the digesting sludge (organic solids generated in the fish system) as the organic carbon source (sludge or pre-denitrification treatment systems) or adding an exogenous electron donor such as methanol (post-denitrification treatment systems). The distinction between the two technologies is based on whether or not an electron donor such as methanol is added. Sludge denitrification uses the endogenous carbon liberated during digestion of organic material in the influent wastewater as the source of organic electron donors while post-denitrification systems (called tertiary treatment in the wastewater industry) require the addition of an exogenous electron donor.

There are three process streams in an aquaculture recirculation system that could be used as influent waste streams to a denitrification unit operation:

1. The high solids loaded center drain discharge could be treated in a two-stage pre-denitrification treatment system, utilizing the fish waste solids as a source of exogenous electron donors (carbon) in an anoxic first stage, followed by a nitrification aerobic second stage bioreactor with recycling of the nitrate-nitrogen rich water back to the first stage.
2. The sidewall discharge or return flow could be treated in a post-denitrification treatment process using an external source of carbon, such as the newer agriculturally derived MicroC™ or Polyhydroxyalkanotes (PHAs) as the electron donor, followed by a small re-aeration reactor before returning to the production tank. PHAs are a family of bioplastic polymers, produced from sugar fermentation.
3. Depending upon discharge requirements, the sludge discharge from the microscreen filters or settling basins could require treatment to remove the high nitrate-nitrogen concentration before discharge, using a single stage pre-denitrification system.

Figure 9.1 shows how a traditional wastewater treatment facility would treat the high VSS, BOD and TAN influent waste stream starting first with solids removal (primary treatment), secondly with an aerobic reactor for BOD oxygenation and nitrification of the ammonia-nitrogen to nitrate-nitrogen (secondary treatment), and finally via an anoxic reactor to denitrify the nitrate-nitrogen to nitrogen gas (tertiary treatment). This is basically the same treatment process that is employed in recirculating aquaculture systems, i.e. solids capture, followed by biofiltration and aeration and when needed denitrification. The major differences are of course the level of TAN and VSS in the influent and effluent, easily an order of magnitude lower in aquaculture systems. In contrast, the nitrate-nitrogen levels are more on parity, with both systems having to meet the same discharge requirements of less than 10 mg/L NO_3-N.

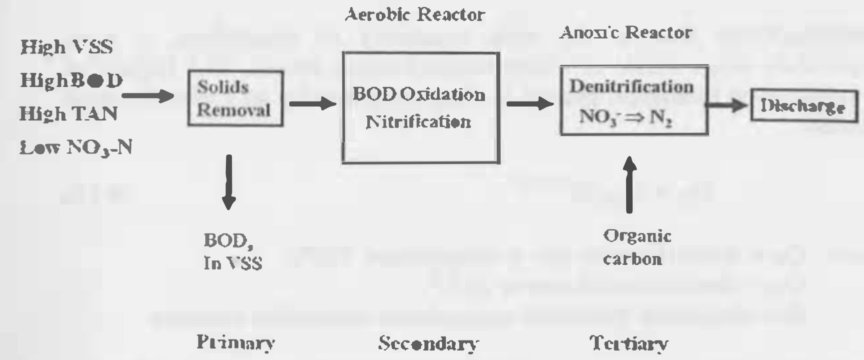

Figure 9.1. A traditional wastewater treatment system with primary solids removal, secondary BOD and nitrogen removal, and finally tertiary denitrification supplemented with an external carbon source.

THE SLUDGE OR PRE-DENITRIFICATION PROCESS

A sludge or pre-denitrification process involves using the endogenous carbon liberated during digestion of organic material in the influent wastewater stream as the source of exogenous electron donors to drive denitrification. Thus, no additional chemicals are required. The two basic strategies in wastewater treatment use either an initial aerobic bioreactor followed by an anoxic denitrification reactor, or an initial anoxic denitrification reactor followed by an aerobic BOD oxidation and nitrification reactor.

Since the water in a RAS system is recirculated through a production tank, the two components can be separated or linked together based on the design strategy. For example, the wastewater stream from the solids capture device (polybead filter, microscreen filter, radial flow clarifier) could be pumped into an anoxic denitrification reactor with a long hydraulic retention time (long enough to drive the water to an anoxic condition). In this sludge tank, denitrification would occur using the endogenous carbon liberated during digestion of organic material in the sludge as the source of organic electron donors. Shnel et al. (2002) reported on just such a strategy where the discharge from a microscreen filter and organic solids that accumulated at the bottom of the fish tanks were diverted into a sedimentation/digestion basin (12 m^3) in a zero-

discharge tilapia production system with a production of 4,868 kg per year. Effluent water from the sedimentation/digestion basin was returned to a point just before the microscreen filter. A trickling tower was used as an "aerobic BOD oxidation and nitrification reactor". Daily removal rates for the sedimentation/digestion basin were equal to 1,689 g NO_3-N per day (see Table 9.3 for a summary of daily removal rates from various systems).

THE TERTIARY OR POST-DENITRIFICATION PROCESS

A tertiary or Post-Denitrification process is employed when the water to be treated contains nitrate-nitrogen and nitrite-nitrogen, but little or no electron donors, i.e. low BOD, low VSS, low dissolved organic matter. This is the more common case in aquaculture where the water being treated is sourced from either the fish tank sidewall discharge in a dual drain system or the return from the water treatment process, i.e., after solids capture-biofiltration-aeration. Here, an organic electron donor is supplied for the denitrification and to accelerate the denitrification process. Traditionally in the wastewater treatment industry, methanol has been used, not because it is better than other choices, but is just cheaper and more readily available. Other traditional organic compounds that have been used or investigated by researchers include ethanol, acetic acid, starch, sugars, carbohydrates and several newer agricultural derived products. In addition, several inorganic electron donors can be used to drive autotrophic denitrification such as hydrogen gas and elemental sulfur.

In the application of post-denitrification processes in aquaculture, biofilm processes are one of the easiest applications to employ and usually preferred since they lack the difficulties encountered in controlling the sludge inventory due to the very low sludge yield during denitrification. There are several fixed film processes that have been utilized in the wastewater treatment industry that could be used in aquaculture including moving bed bioreactors, fluidized sand beds, submerged filters and several types of sand filters. One of primary advantages of these types of filters is the high denitrification rate in relation to the required volume of the biofilter. For example, the maximum daily denitrification rate for triplicate submerged filters using methanol, starch, acetic acid and molasses as carbon sources was 670 to 680 g NO_3-N/m^3-day (Hamlin et al., 2008) in a commercial aquaculture facility.

9.3 Factors Controlling Denitrification

When considering the various factors known to control denitrification, it is important to realize that reduction of nitrate to nitrogen gas proceeds via various intermediates, among them nitrite, a compound extremely toxic to aquatic animals. In aquaculture systems, it is important to identify not only the factors that inhibit or enhance denitrification, but also those that cause intermediate nitrite accumulation by denitrifiers.

Oxygen. Denitrifying bacteria and microbial populations have built in control systems that ensure that the most efficient forms of energy generation are utilized (EPA, 1993). In the case of denitrifying bacteria, dissolved oxygen is the preferred choice for the bacteria's oxygen source over nitrate or nitrite. Thus under aerobic conditions, denitrification is not expected to take place. However, in aquaculture systems as well as other water treatment systems, nitrate removal in apparently aerobic environments such as nitrifying filters is not uncommon. The observed nitrate removal under these conditions is due to the heterogeneity of the environment.

Accumulation of organic matter in an aerobic environment may lead to anoxic conditions within the organic layer or biofilm and may provide suitable conditions for proliferation and activity of denitrifiers. Furthermore, short anaerobic periods of normally aerobic environments may lead to considerable nitrate losses. This is not surprising when considering the fact that denitrifiers form an intrinsic part of microbial communities developing under aerobic conditions. Low oxygen concentrations in the environment may lead to nitrite accumulation as a result of the differential repression of nitrite reductase synthesis and activity as compared to nitrate reductase (Komer and Zumft, 1989; Coyne and Tiedje, 1990).

PH In general, denitrification is much less sensitive to pH than nitrification (EPA, 1993). Denitrification can occur over a wide range of pH values, with optimal pH values between pH 7 and 8. Some observations suggest that the denitrification rates are depressed above and below pH of 6.0 and 8.0. In cases where high concentrations of nitrate are to be removed, the resulting production of alkalinity can increase the pH of the medium. Shifts in the pH values can lead to the accumulation of intermediate products, such as NO_2^-, NO_2 and N_2O.

Temperature Due to the high versatility of denitrifiers, a wide temperature range exists at which denitrification occurs. The impact of temperature on biological system is often described by an Arrhenius-type function:

$$Q_T = Q_{20}\, \Theta^{(T-20)} \qquad (9.15)$$

where Q_T = denitrification rate at temperature T(C°)
Q_{20} = denitrification rate at 20 C°
Θ = simplified Arrhenius temperature-dependent constant

Within the range of 10 to 30°C, temperature activity coefficients generally fall within the range of 1.08 to 1.20 (Metcalf and Eddy, 1991) and corresponding Q_{10} values within the range of 2-3 (Heinen, 2006). Although the above function is useful for modeling denitrification, it is limited to a defined temperature range and the value of Θ is often site specific (EPA, 1993).

Salinity. Based on the few studies conducted, no clear consensus exists on the effect of salinity on denitrification (Glass and Silverstein, 1999). Denitrification is an important nitrogen transformation process in some marine systems, including marine recirculating systems (Gelfand et al., 2003). No significant differences were found in denitrification rates between freshwater and marine recirculating systems by van Rijn et al. (2006). The adaptation of denitrifiers from fresh to seawater was examined by Park et al. (2001), who found that temporary nitrite accumulation occurred during the acclimation period. Upon completion of this period, no differences in denitrification rates were observed between fresh and seawater environments.

Inhibitors. Denitrifiers are much less sensitive to inhibitory compounds than are nitrifiers (EPA, 1993). In general, inhibitors such as ethanol or phenols would be expected to have a similar degree of impact on denitrification and heterotrophic aerobic respiration. Thus, the nitrifiers in a system will need time to adjust to the low level inhibitors that are used to support the denitrification process. With the understanding that many bacteria and microbial systems have the ability to acclimate to higher levels of inhibitory compounds, if any such compounds are used, they should be used very cautiously and discontinued or reduced if any inhibitory response is observed.

CARBON SOURCE. Heterotrophic denitrifiers obtain their electrons and protons required for reduction of nitrate to elemental nitrogen from organic carbon compounds. Almost any compound that is degraded with oxygen as the electron acceptor will also serve as an electron donor with nitrate. As a result, the list of organic compounds that can serve as organic substrate for denitrification is especially long. Such compounds include carbohydrates, organic alcohols such as methanol and ethanol, amino acids, acetic acid, fatty acids and organics in the wastewater.

The C/N ratio required for complete nitrate reduction to nitrogen gas by denitrifying bacteria depends on the nature of the carbon source and the bacterial species (Payne, 1973). For most readily available organic carbon sources g COD/g NO_3^- ratios in the range from 3.0 to 6.0 enable complete reduction of nitrate to elemental nitrogen (Montieth et al., 1979; Narcis et al., 1979; Skrinde and Bhagat, 1982). Also denitrification rates are to a large extent dictated by the type of carbon source. In wastewater treatment plants as well as in aquaculture systems, exogenous carbon substrates are often used to drive the denitrification process with methanol being most widely used (Payne, 1973). This latter carbon source is relatively cheap and causes the development of a distinct microbial population and, hence, a predictable performance of a reactor. It should be noted, however, that in reactors fed with specific organic compounds, the resulting denitrifying population is often far from predictable. Other forms of organic matter present in the reactor, such as those liberated by the decay of microorganisms or present in the water to be treated, may give rise to a diverse microbial population. In this context it is worth mentioning that endogenous carbon, liberated by the digestion of organic sludge and manures in recirculating systems, forms an excellent carbon source for denitrifiers and gives rise to a highly versatile denitrifying community (Aboutboul et al., 1995; Sich and van Rijn, 1997; Cytryn et al., 2003).

Carbon limitation will result in the accumulation of intermediate products of denitrification such as NO_2 and N_2O, while excess carbon will promote dissimilatory nitrate reduction to ammonium (Tiedje, 1990). The type of carbon source also affects the level of nitrite accumulation in denitrifying reactors (McCarthy et al., 1969). Certain carbon sources may lead to a specific enrichment of nitrite-accumulating bacteria, as was found in a study on denitrifying reactors where addition of fermentable substrates enhanced the growth of such bacteria (Wilderer et al., 1987). Alternatively, denitrifiers, capable of complete reduction of nitrate to elemental nitrogen gas, may accumulate nitrite when grown on certain carbon sources (Blaszczyk, 1993; Nishimura et al., 1979; 1980; van Rijn et al. 1996). Temporary carbon starvation leads to nitrite accumulation in denitrifying bacteria with constitutive nitrate reductases and inducible nitrite reductases (Barak, 1997). Competition between nitrate and nitrite reductases for common electron donors is an additional factor causing nitrite accumulation by some denitrifiers (Betlach and Tiedje, 1981; Kucera et al., 1983; Thomsen et al., 1994).

Potential carbon sources for passive denitrification are polyhydroxyalkanotes (PHAs). PHAs are a family of bioplastic polymers, produced from sugar fermentation. The biodegradation of PHA's in the presence of nutrients releases organic carbon, which makes them an ideal substrate for a self-regulating, passive denitrification reactor. PHAs offer a potential low maintenance, cost effective method to achieve denitrification, since they act as both an organic carbon source and substrate for denitrifying bacteria to attach to (Boley et al., 2000; Ebeling & Drennan, 2006). This in turn, eliminates the need for sophisticated control systems and handling of hazardous chemicals, required by the conventional methods of treatment (Boley et al., 2000, Lee et al., 2000). Several studies have demonstrated that the PHAs are a rich source of carbon with denitrification rates in simple fixed bed reactors of approximately 2 kg/m^3 media-d. One of the major problems encountered with earlier studies was excessive biofloc formation produced by the heterotrophic bacteria (Gutierrex-Wing et al., 2006). The successful results from these initial studies led to the hypothesis that the problems associated with clogging and short circuiting of the PHA filter bed could be addressed by utilizing a PolyGeyser® bioclarifier (bead filter) to promote a healthy thin biofilm via its characteristic frequent, gentle backwashing, which eliminates the clogging problem observed with packed beds (Ebeling & Drennan &, 2006).

Another alternative carbon-source for biological denitrification is MicroC™. MicroC™ is a proprietary wastewater treatment chemical developed by Environmental Operating Solutions, and designed specifically as a viable, non flammable, agriculturally derived electron donor. While this alternative has not been applied to an aquaculture system yet, studies (Cherchi, et al., 2008) have demonstrated that MicroC™ can effectively support denitrification and showed a slightly better performance compared to methanol, especially when used in a post denitrification process and under low temperature conditions.

LIGHT. In a study on a denitrifying bacterium isolated from a denitrifying fluidized bed reactor in a recirculating aquaculture facility, nitrite accumulation resulted from light exposure. It was shown that at light intensities as low as 5% of full sunlight intensity, nitrite

accumulated as a result of light inhibition of cytochromes involved in nitrite reduction in this bacterium (Barak et al., 1998).

NITRATE. Nitrate reduction follows classical Michaelis-Menten kinetics (Michaelis and Menten, 1913) with respect to the nitrate concentration in the medium. Since affinity values for nitrate by denitrifiers are in the low μmolar range (Betlach and Tiedje, 1981), nitrate removal in most environments, including reciculating systems, can be perceived as a zero-order reaction.

9.4 EFFECT OF DENITRIFICATION ON ALKALINITY

Stabilization of the buffering capacity of culture water by denitrification is considered an additional factor favoring the use of denitrification in recirculating systems. The amount of acid required to titrate the bases in water is a measure of the alkalinity of water. A chemical reaction where acid is produced will lower the alkalinity of the water, while the opposite holds for a reaction in which acid is consumed or hydroxyl ions are produced. Weihrauch et al. (2009) provide a review of the literature involving ammonia excretion by fish and aquatic crustaceans. Although, not totally conclusive, they concluded that the dominant form of ammonia being excreted by fish is NH_3 as opposed to ammonium, NH_4^+. Recent research by Timmons and Ebeling (manuscript in preparation) has verified based upon measurement of alkalinity loss that fish do excrete ammonia primarily as NH_3, which means that the combination of ammonia excretion and nitrification will consume on a net basis approximately 1 unit of alkalinity (1 equivalent per mole of ammonia excreted), not 2 units as has been previously reported in many texts. The denitrification process will produce one unit of alkalinity per mole of nitrate denitrified, so the net overall process means that system alkalinity would remain unchanged. This is a very important point for overall system stability of water quality in an aquaculture system.

As ammonia can react with H^+, each mole of ammonia excreted by fish raises the alkalinity by 1 equivalent (eq). Within the normal pH range of aquaculture systems, ammonia is protonated to NH_4^+ (Eqs. 9.16a and 9.16b) once it enters the water.

It can be seen from equations 9.16 that the alkalinity, lost by conversion of NH_3 to NH_4^+, is recaptured by the formation of OH^-, an ion which also reacts with H^+.

$$NH_3 + H_2O \leftrightarrow NH_4^+ + OH^- \qquad (9.16a)$$

$$NH_3 + H_2O = NH_4OH = NH_4^+ + OH^- \qquad (9.16b)$$

(Alkalinity increase = 1 eq of alkalinity per mole excreted NH_3)

During nitrification of ammonium (NH_4^+), alkalinity decreases by 7.14 g $CaCO_3$ (2 eq) for each g of ammonia oxidized to nitrate according to the following simplified stoichiometry of the reaction (Eq. 9.17)

$$NH_4^+ + 2O_2 = NO_3^- + 2H^+ + H_2O \qquad (9.17)$$

(Alkalinity loss = 2 eq of alkalinity per mole NH_4^+ or 7.14 mg $CaCO_3$/mgTAN)

Thus, the alkalinity loss through NH_3 excretion and subsequent nitrification is 1 eq per mole of NH_3 excreted by the fish. This alkalinity loss is compensated for when in addition to nitrification, denitrification is used as a water treatment stage. Heterotrophic denitrification causes a release of hydroxyl ions and hence raises alkalinity. It can be estimated that each g of nitrate-N reduced to N_2 causes an alkalinity increase of 3.57 g $CaCO_3$ according to the following stoichiometry (Eq. 9.18):

$$2NO_3^- + 12H^+ + 10\,e^- = N_2 + 6H_2O \qquad (9.18)$$

(Alkalinity gain = 1 eq of alkalinity per mole of NO_3 or 3.57 g $CaCO_3$/g NO_3^--N)

Autotrophic denitrification can be applied for nitrate removal in some recirculating systems (Tal and Schreier, 2004). With sulfide as electron donor, alkalinity is gained through two separate processes. In the first process (Eq. 9.19), sulfate is reduced to sulfide. The latter compound is reoxidized to sulfate with concomitant reduction of nitrate (Eq. 9.20) in the second process.

$$SO_4^{2-} + 10H^+ + 8e^- = H_2S + 4H_2O \qquad (9.19)$$

(Alkalinity gain = 2 eq of alkalinity per mole SO_4^{2-} or 100 g $CaCO_3$/mole SO_4^{2-})

$$5H_2S + 8NO_3^- \rightarrow 5SO_4^{2-} + 4N_2 + 4H_2O + 2H^+ \qquad (9.20)$$

(Alkalinity loss = 2 eq per 5 moles H_2S or 20 g $CaCO_3$/mole HS^-)

It follows from equations 9.18 and 9.19 that the coupled processes of sulfate reduction, sulfide oxidation and nitrate reduction result in a net alkalinity generation of 400 g $CaCO_3$ per 8 moles of nitrate reduced or 3.57 mg $CaCO_3$ per mg of nitrate-N reduced.

It is unlikely, however, that autotrophic denitrification will occur in the presence of degradable organic matter (the normal case in RAS) since heterotrophic denitrification will dominate here. Where this condition might occur, though, is downstream of the primary denitrification reactor (Eq. 9.17) where BOD has been consumed and then treatment of the effluent by a microscreen filter that would remove most of any remaining organic matter in the flow stream. Autotropic denitrification was also found to play an important role in heterogeneous, sludge denitrification reactors (Cytryn *et al.*, 2005a, 2005b, 2006; Gelfand *et al.*, 2003, Neori *et al.*, 2007).

"Rule of Thumb"

Fish contribute 3.57 g of alkalinity as $CaCO_3$ per g of NH_3-N excreted

For each gram of NH_3-N converted to N_2 via nitrification and denitrification, 3.57 grams of alkalinity as $CaCO_3$ are destroyed

Net change in alkalinity = zero

9.5 Effect of Denitrification on Phosphate Removal

Enhanced biological phosphorus removal (EBPR) from domestic wastewater in activated sludge plants is accomplished by alternate stages, where sludge is subjected to anaerobic and aerobic conditions. Phosphorus is released from bacterial biomass in the anaerobic stage and is assimilated by these bacteria in excess as polyphosphate (poly-P) during the aerobic stage. Phosphorus is removed from the process stream by harvesting a fraction of the phosphorus-rich bacterial biomass

(Toerien *et al.*, 1990). Some of these polyphosphate accumulating organisms (PAO) also are capable of poly-P accumulation under denitrifying conditions, i.e. with nitrate instead of oxygen serving as the terminal electron acceptor (Barker and Dold, 1996; Mino *et al.*, 1998). Studies on poly-P accumulating organisms have revealed the involvement of specific metabolic properties under anaerobic, aerobic and anoxic conditions (Mino *et al.*, 1998). Under anaerobic conditions, acetate or other low molecular weight organic compounds are converted to polyhydroxyalkanoates (PHA), poly-P and glycogen are degraded and phosphate is released. Under aerobic and anoxic conditions, PHA is converted to glycogen, phosphate is taken up and poly-P is synthesized intracellularly. Under the latter conditions, growth and phosphate uptake is regulated by the energy released from the breakdown of PHA.

Some heterotrophic denitrifiers exhibit phosphorus storage in excess of their metabolic requirements through poly-P synthesis under either aerobic or anoxic conditions, without the need for alternating anaerobic/aerobic switches (Barak and van Rijn, 2000a). Unlike PAO, these denitrifiers were unable to use PHA as an energy source for poly-P synthesis and derived energy from oxidation of external carbon sources. The feasibility of this type of phosphate removal was demonstrated for freshwater as well as marine recirculating systems (Barak and van Rijn, 2000b; Shnel *et al.*, 2002; Barak *et al.*, 2003; Gelfand *et al.*, 2003; Neori *et al.*, 2006). In the culture water of these systems, stable orthophosphate concentrations were found throughout the culture period. Phosphorus immobilization took place in the anoxic treatment stages of the system where it accumulated to up to 19% of the sludge dry weight.

9.6 Effluent versus On-line Treatment

Nitrate may accumulate to high concentrations in recirculating systems in which water treatment is based on removal of organic matter and nitrification. Daily water exchange is often the method of choice to avoid its accumulation to harmful concentrations (Masser et al., 1999). Due to more stringent regulations in most countries, it is expected that nitrate removal from aquaculture effluents will be required in the near future. The removal of nitrate from effluents involves the reduction from relatively high concentrations to relatively low, permissible nitrate concentrations. Regulatory allowable nitrate concentrations in the effluent are well above reaction rate limiting concentrations and as such denitrification during effluent treatment is not limited by nitrate

concentrations. However, whether or not nitrate is rate limited depends to a large extent on the homogeneity of the denitrification reactor. Under typical heterogeneous conditions, it is probable that some regions within the reactor will be nitrate depleted. Therefore, reactors operated at higher ambient nitrate concentrations may be more efficient in nitrate removal than reactors operated at low ambient nitrate concentrations. Securing high nitrate concentrations in the denitrification treatment step can be accomplished by on-line treatment of nitrate within the recirculating systems. Due to the nitrate tolerance of most cultured organisms, aquaculture systems with on-line nitrate treatment units can be operated at ambient nitrate concentrations, which may be one order of a magnitude higher than those in systems where effluent water is treated.

9.7 TYPES OF REACTORS

In treatment of wastewater several denitrifying reactors are used. Reactors can be divided in suspended growth systems and attached growth systems. Suspended growth systems are either completely-mixed or plug-flow reactors where under anoxic and carbon-rich conditions a denitrifying biomass develops. Similar to activated sludge treatment, part of the sludge removed from the reactors is recycled in order to maintain a stable biomass of denitrifying organisms. In some recirculating systems where such reactors are applied, it was found that there was no need for sludge removal (see below). The upflow anaerobic sludge-blanket (UASB) process is a special type of suspended growth system. In this reactor, wastewater flows upward through a sludge blanket composed of biologically formed granules. This type of reactor, mainly used for anaerobic digestion of organic matter, is also used for nitrate removal in some wastewater treatment as well as aquaculture plants. Bioreactors where the bacteria is attached to a surface are called fixed-film or packed bed reactors, including fluidized bed reactors, and, more recently, moving bed reactors[2] (Odegaard, 2006).

[2] Note by Authors: A moving bed would need to use a mechanical stirrer and not an aeration system to maintain anoxic conditions during the mixing process.

Table 9.1. Denitrification Reactors in Recirculating Systems

Denitrifying reactor	Organism cultured	Carbon/electron donor	Reference
Freshwater systems			
Activated sludge	Carp	Endogenous	Meske (1976)
Activated sludge	Tilapia, Eel	Glucose/ methanol	Otte and Rosenthal (1979)
Activated sludge	Trout	Hydrolyzed corn starch	Kaiser and Schmitz (1988)
Digestion basin and fluidized bed reactor	Tilapia	Endogenous	van Rijn and Rivera (1990); Arbiv and van Rijn (1995); Shnel et al. (2002)
Activated sludge	Eel	Endogenous	Knosche (1994)
Packed bed reactor	Not specified	Methanol	Abeysinghe et al. (1996)
Packed bed reactor	Not specified	Endogenous	Phillips and Love (1998)
Polymers	Ornamental carp	Endogenous	Nagadomi et al. (1999)
Polymers	Ornamental fish	Biodegradable polymers	Boley et al. (2000)
Packed bed reactor	Eel	Methanol	Suzuki et al. (2003)

Table 9.1 (cont.) Denitrification Reactors in Recirculating Systems

Denitrifying reactor	Organism cultured	Carbon/electron donor	Reference
Marine Systems			
Packed bed reactor	Atlantic and Chinook salmon	Methanol	Balderston and Sieburth (1976)
Packed bed reactor	Japanese Flounder	Glucose	Honda et al. (1993)
Packed bed reactor	Squids	Methanol	Whitson et al. (1993)
Packed bed reactor	Not specified	Ethanol	Sauthier et al. (1998)
Fluidized bed reactor	Ornamental fish	Methanol	Grguric and Coston (1998)
Polymers	Not specified	Glucose	Park et al. (2000)
Packed bed reactor	Ornamental fish	Methanol	Grguric et al. (2000a,b)
Packed bed reactor	Shrimp	Ethanol/ Methanol	Menasveta et al. (2001)
Polymers	Ornamental fish	Starch	Tal et al. (2003a)
Digestion basin and fluidized bed reactor	Gilthead seabream	Endogenous	Gelfand et al. (2003)
Moving bed reactor	Gilthead seabream	Starch	Morrison et al. (2004)
	Ornamental	Methanol	Labelle et al. (2005)
	Ornamental	Methanol	Dupla et al. (2006)

9.8 MBBR PROCESSES FOR DENITRIFICATION

The moving bed bioreactor (MBBR) was developed in Norway in the early 1980's as a means for retrofitting existing wastewater treatment facilities in a cost effective manner to remove nitrogen through either a pre-denitrification or post-denitrification processes. A significant advantage of the MBBR in upgrading existing wastewater treatment plants was its small footprint and low maintenance in comparison to the operational and maintenance issues associated with trickling filters and activated sludge systems. MBBR technology is currently widely used in European wastewater treatment facilities and in both small and large-scale commercial aquaculture operations for nitrification.

The MBBR is an attached growth biological treatment process based on a continuously operating, non-clogging biofilm reactor with low head loss, a high specific biofilm surface area, and no requirement for backwashing. The bacterial biomass grows on the media carriers and moves freely in the water volume of the reactor. The reactor can be operated under either aerobic conditions for nitrification or anoxic conditions for denitrification. For nitrification, the media is maintained in constant circulation via a course air bubble aeration system creating aerobic conditions and for denitrification via a submerged mixer (normally a horizontal shaft mounted banana mixer) to promote anoxic or anaerobic conditions. Media usually occupies up to 67% of the reactor volume (normally 50% fill), in that at higher percentage fill reduces mixing efficiency. The media is kept within the reactor volume by an outlet sieve or screen, which may be vertically mounted, rectangular mesh sieves, or cylindrical bar sieves, vertically or horizontally mounded. The media most often used (Kaldnes K1) is made of high density polyethylene (density 0.95 g/cm^3) and shaped as a small cylinder with a cross on the inside of the cylinder and 'fins' on the outside (Ødegaard et al., 2004). Other media has also been used, although all have the characteristic of a protected area for biofilm growth.

Figure 9.2 Anoxic reactor with horizontally mounted shaft mixers and rectangular mesh sieves and with aeration system, operated either as an aerobic or anoxic reactor (Ødegaard et al, 2004).

Agitation within the reactor maintains the media in constant motion creating a scrubbing effect that prevents clogging and sloughs off excess biomass. Since MBBR's are an attached growth process, treatment capacity is a function of the specific surface area of the media. This is often reported as the specific surface area of the reactor, equal to the total surface area of the media divided by the volume of the reactor, or the media specific surface area multiplied by the fraction of the total reactor volume that the media occupies. In some cases, the total surface area of the media that is available for biofilm development divided by the volume of the reactor is used, reflecting the significant abrasion of biofilm off the outer surface of some media types. For Kaldnes K1 media, the specific biofilm surface area is 500 m^2/m^3 , so for vessels that are 50% filled with media, the surface area per unit volume of vessel would be: 250 m^2/m^3 and at 67% fill: 350 m^2/m^3.

The MBBR technology has significant potential for denitrification in commercial recirculating aquaculture systems as seen by its increased use as a nitrifying biofilter. The MBBR technology can be used for nitrogen removal by pre-denitrification, post-denitrification or some combination of the two processes (see Section 9.2 for descriptions). The limitations of the pre-denitrification process is the high recycle rate of oxygen enriched water and the potential limiting effect due to insufficient carbon availability in the influent water. In a post-denitrification process, high denitrification rates are possible with the addition of easily biodegradable carbon sources. For example, data from a MBBR pilot plant in Norway demonstrated a maximum denitrification rate of 2.5 g N/m^2-d using ethanol and 2.0 g N/m^2-d for methanol at 16 °C (Aspegren et al, 1998). Assuming a specific area effective for biofilm growth of 500 m^2/m^3 for the carrier media, yields a volumetric denitrification rate of 1.23 kg N/m^3*d for ethanol and 1.00 kg N/m^3*d for methanol. Although lower than the rates for a more traditional fluidized bed or a submerged filter, the MBBR process provides for an easy and a more flexible operation than these other processes. As compared to activated sludge reactors for example, there is no need for sludge recirculation. In addition, almost any bioreactor shape can be used and different operating loadings in a given reactor volume can be chosen by changing the media filling fraction (Ødegaard, 2006).

Design Parameters for MBBR Denitrification

Ødegaard et al. (2004) reviewed the use in Norway of the MBBR in the wastewater treatment process for BOD/COD removal, nitrification and denitrification. The results from three Norwegian treatment plants were presented that used both a pre-denitrification and a post-denitrification process. Each of the facilities consisted of either seven or nine treatment reactors in series. The first reactor was anoxic, with only mixers and no aeration for pre-denitrification. The next reactor was equipped with both mixers and aeration to allow flexibility in operation. The middle reactors were operated aerobically for nitrification and equipped with aeration diffuser grids. The second from the last had both aeration and mixers, but was run mostly with mixing and little aeration. This was to consume as much oxygen as possible to provide a low dissolved oxygen waste stream for recycling back to the first pre-denitrification anoxic reactor in the series. Finally, the last reactor was equipped with mixers only and a source of carbon for post-denitrification. Lastly in the series was a small aeration chamber to re-aerate the water before discharge.

What is most important about the above design example was that the water temperature ranged from 6 to 16 °C. Although this was a wastewater treatment process, the performance of the process was very similar to what an aquaculture system would be required to accomplish. In this case, the incoming total nitrogen levels to the final post-denitrification reactor ranged from 16 to 48 mg-N/L and the effluent from 2.9 to 12.7 mg-N/L. At the average influent concentration of 27 mg/L, the removal rate was 76% with an ethanol consumption of 1.48 kg ethanol/kg N (3.1 kg COD/kg $N_{removed}$). The denitrification rate was estimated to be 2 mg NO_3-N/m^2-d, using methanol at a temperature of 15 °C. Almost all of the nitrogen was removed by the post-denitrification reactor.

9.9 DESIGN OF DENITRIFICATION REACTORS

An effective denitrification reactor can be designed by considering the following parameters:

- Nitrate-nitrogen production in the system
- The maximum allowable nitrate concentrations in the culture water
- The volume of reactor required to match the nitrate load
- Retention time of the reactor
- The type of electron donor (organic or inorganic) and its stoichiometric requirements in the denitrification process
- Oxygen concentrations in the reactor

Explanations & Assumptions

- The TAN load of the system has been described in details in the previous Biofiltration Chapter 7 and is generally assumed to be ~3% of the daily feeding rate (9.2% of the Protein content of feed). Using a conservative approach, it may be assumed that TAN levels in the culture water are kept low (< 2 mg/L) and that all TAN is oxidized to nitrate in the nitrifying filter. For practical purposes, it is further assumed that of the generated nitrate, some fraction is removed by daily water exchange of the system (See Chapter 3: Mass Balances) and the remainder by induced denitrification.
- Ambient nitrate levels are set at concentrations after considering the nitrate tolerance of the cultured organism and the allowable nitrate concentrations in the discharge effluent of the recirculating system. It should be noted that, especially in marine systems, operation of the system at low ambient nitrate concentrations *carries the risk* of sulfide formation in anaerobic, nitrate-poor parts of the system. In general, semi-closed recirculating systems require operation at lower ambient nitrate levels than closed systems since nitrate concentrations tolerated by most aquacultured organisms are significantly higher than those permitted in the effluent water.
- The volume of the denitrification reactor is based on the denitrification rate within the various filters, which in turn depends on the type of reactor and type of electron donor (see Tables 9.1 and 9.3). Since, nitrate reduction will be optimal with non-limiting nitrate concentrations throughout the reactor (see above), smaller reactors are required when

systems are operated at relatively high ambient nitrate concentrations. It should be noted that just as for nitrification (see Chapter 7: Biofiltration); the total daily mass of nitrate removal defines the effectiveness of the filter rather than the efficiency of nitrate removal during a single pass, i.e., what is important is the product of change in concentration across the filter and flow rate.
- The required ambient nitrate concentrations in the system are the main determinant for the retention time at which the denitrification reactors are operated. When the reactors are operated at too short retention times, maintenance of oxygen-free conditions within the reactor might be difficult. Conversely, too long retention times might cause the production of harmful anaerobic metabolites in the reactor, e.g., sulfide.
- Denitrification rates are greatly influenced by the type of electron donor. The stoichiometric relationship between nitrate and common organic and inorganic electron donors are presented in Table 9.2. These stoichiometric equations, however, should be consulted with care as they might divert significantly from the *in situ* relation between electron/carbon donor requirements within the denitrification reactor. Main factors underlying this difference are the consumption of the electron donor by other biological processes in the reactor, e.g. aerobic respiration, and the differences in carbon conversion into bacterial biomass for maintenance and growth. In the case where external carbon sources are used to fuel denitrification, one should be aware of the fact that treatment water in recirculating systems contains organic matter. It is inevitable, therefore, that even in reactors operated with a well-defined external carbon source, the *in-situ* denitrification rates are different from what would be expected based on theoretical assumptions. Hence, system-specific trials for determining the relation between electron/carbon donor and nitrate are required.
- Oxygen removal from the influent water or within the denitrification reactor is essential to secure denitrifying activity. One option is the removal of oxygen by purging the influent water with nitrogen gas. Especially, in reactors operated with the addition of an external carbon source, such active removal of oxygen is often the only option for securing anoxic conditions within the reactor. Oxygen consumption within the reactor may also lead to anoxic conditions within the reactor. Especially in reactors where denitrification is fueled by endogenous carbon sources, oxygen consumption is rapid and active oxygen removal from the influent water is not required. It should be noted, however, that under such conditions, part of the organic carbon is aerobically respired and unavailable for denitrification.

9.10 Design Example

As described in earlier chapters, Omega Industries (OI) requires an engineering plan for the construction and operation of an Omega Fish Aquaculture Facility (OFAF). The production strategy is a three stage system: juvenile, fingerling and growout stages. Although the Omega Fish is very hardy and can handle high nitrate levels, regulatory demands and marketing considerations require us to maintain the tank and the discharge water nitrate concentrations from the system as low as possible, e.g., < 10 mg/L nitrate-N. At this stage, a sophisticated engineering design for the state-of-the-art denitrification system would be in order. The difficulty is that at the current stage in the development of denitrifying systems, the design guidelines for such a system do not exist or are still in the research stages. Although numerous laboratory scale systems have been designed at research universities, very few are available as turn-key commercial systems. The design presented below is based on the success of the MBBR for nitrification in commercial aquaculture systems, and the success of MBBR for denitrification in Europe.

A possible design for a post-denitrification process using a MBBR for the proposed denitrification system could consist of the following:

- *Nitrate-nitrogen production in the system*

For this design example, a juvenile/growout pod consisting of two juvenile tanks and two growout tanks will be used (see Chapter 5 Solids Capture for previous presented design of the fish tank and pod design). The maximum design biomass for this POD was 2267 kg and the daily maximum feed rate was estimated at 27.5 kg. Assuming a high quality feed with a 38% protein content (0.035 kg TAN/kg feed), suggests a daily production of 0.96 kg NH_3-N/day. It is difficult to determine the passive denitrification that can occur in any production system, so we will make that a safety factor in the design and assume it is zero. Thus if we assume that all of the TAN is converted into nitrate-nitrogen in the biofilter and that an insignificant quantity is lost in the discharge flow from the system, then 0.96 kg NO_3-N is generated per day.

- *The maximum allowable nitrate concentrations in the culture water*

This is a really simple design, so the nitrate-nitrogen will be maintained as low as possible in the tank and in the discharge by over sizing the removal system. Note that if the denitrification rate is equal to the production rate of nitrate (goal of design), then the equilibrium concentration of nitrate in the

fish system will approach zero if the flow through component of water (the aquaculture system water discharge rate) being added is free of nitrate.

- *The volume of reactor required to match the nitrate load*

Volumetric denitrification removal rates (VDR) have been determined for several different types of MBBR media and are on the order of 2 g N/m²*d. Thus the required surface area would be:

$$Surface\ Area = \frac{m_{NO_3-N}}{VDR} = \frac{\left(963\,\dfrac{g\ NO_3-N}{day}\right)}{\left(2\,\dfrac{g\ NO_3-N}{m^2\cdot day}\right)} = 481\,m^2\,(5180\,ft^2)$$

The effective biofilm surface area for a commonly used MBBR media, i.e. K1 Kaldnes media is 500 m^2/m^3. Thus the volume of media required is:

$$Volume = \frac{Surface\ Area}{SSA} = \frac{(481\,m^2)}{\left(500\,\dfrac{m^3}{m^2}\right)} = 0.96\,m^3 \quad (34\,ft^3)$$

Assuming a 67% fill factor for the MBBR suggests a Bioreactor volume of 1.43 m^3 (0.96 m^3/0.67) or 378 gallons. Mixing is accomplished with a 1/4 hp horizontal shaft reducing gear box on top of a mounted propeller.

- *Retention time of the reactor*

Based on several municipal wastewater treatment systems, assume a Hydraulic Retention Time (HRT) from 20 to 30 minutes. This would suggest a flow rate through the MBBR of from 10 to 15 gpm.

- *The type of electron donor (organic or inorganic) and its stoichiometric requirements in the denitrification process*

The organic carbon source chosen for this example is an agriculturally derived product: MicroC™ Premium Carbon Sources with a dosing requirement that would have to be field determined but is estimated at a maximum of 5:1 ratio, MicroC™ to nitrate-nitrogen.

Table 9.2. Stoichiometry of heterotrophic and autotrophic denitrification with selected electron/carbon donors*

Substrate	Equation	Organic requirement (g COD/ g NO_3^--N)
Acetate	$0.819CH_3COOH + NO_3^- \rightarrow$ $0.068C_5H_7NO_2 + HCO_3^- + 0.301CO_2 + 0.902H_2O + 0.466N_2$	3.72
Methanol	$1.08CH_3OH + NO_3^- + H^+ \rightarrow$ $0.065C_5H_7O_2N + 0.467N_2 + 0.76CO_2 + 2.44H_2O$	3.70
Ethanol	$0.613C_2H_5OH + NO_3^- \rightarrow$ $0.102C_5H_7NO_2 + 0.714CO_2 + 0.286OH^- + 0.980H_2O + 0.449N_2$	4.19
Glucose	$C_6H_{12}O_6 + 2.8\ NO_3^- + 0.5\ NH_4 + 2.3\ H^+ \rightarrow$ $0.5\ C_5H_7NO_2 + 1.4N_2 + 3.5CO_2 + 6.4H_2O$	4.86
Organic matter from a recirculating system **	$C_7H_{13.4}O_{3.5}P_{0.3}N + 3.3NO_3^- + 2.6H^+ \rightarrow$ $1.6N_2 + 2.9CO_2 + 0.2NH_3 + 4.9H_2O + 0.8C_5H_7O_2P_{0.1}N + 0.2\ PO_4^{3-}$	5.7
Hydrogen	$2NO_3^- + 5H_2 \rightarrow$ $N_2 + 4H_2O + 2OH^-$	
Sulfide	$14NO_3^- + 5FeS_2 + 4H^+ \rightarrow$ $7N_2 + 10SO_4^{2-} + 5Fe_2^+ + 2H_2O$	
Elemental sulfur	$10NO_3^- + 11S^0 + 4.1HCO_3^- + 0.5CO_2 + 1.71NH_4^+ + 2.54H_2O \rightarrow$ $0.92C_5H_7NO_2 + 11SO_4^{2-} + 5.4N_2 + 9.62H^+$	

*** Data compiled from Ahn (2006), Klas et al. (2006) and Mateju et al. (1992)**
**** Assuming a maximum bacterial yield of 0.53 g COD per g COD**

Table 9.3. Volumetric denitrification rates of denitrifying reactors used in aquaculture facilities

Denitrifying reactor	Carrier	Carbon source	Nitrate removal rate (mg NO_3-N/L/hr)	Hydraulic retention time(hr)	Reference
Freshwater systems					
Fluidized bed	Sand	Endogenous	35.8	0.22	Arbiv and van Rijn (1995)
Packed bed	Biodegradable polymers	PHB ($C_4H_6O_2$)n PCL ($C_6H_{10}O_2$)n Bionolle ($C_6H_8O_4$)n	1.5–166	0.75	Boley *et al.* (2000)
Packed bed	Polyethylene	Methanol	1.8*	Not specified	Suzuki *et al.* (2003)
Digestion basin	Sludge	Endogenous	5.9	5.2	Shnel *et al.* (2002)
Fluidized bed	Sand	Endogenous	55.4	0.028	Shnel *et al.* (2002)
Packed bed	Freeze-dried alginate beads	Starch	26.0	0.16	Tal *et al.* (2003a)
Digestion basin	Sludge	Endogenous	1.5	1.19	Gelfand *et al.* (2003)
Packed bed	Polyethylene Media	Methanol Starch Molasses Acetic Acid	28	1.67	Hamlin *et al.* (2008)
Packed bed	Cotton wool	Endogenous + Carrier	3.55 (max)*	2.25	Singer *et al.* (2008)

Table 9.3 (cont.) Volumetric denitrification rates of denitrifying reactors used in aquaculture facilities

Denitrifying reactor	Carrier	Carbon source	Nitrate removal rate (mg NO_3-N/L/hr)	Hydraulic retention time(hr)	Reference
Marine Systems					
Packed bed	Plastic carrier	Glucose	1.7	55	Honda *et al.* (1993)
Packed bed	Brick granules	Ethanol	100	0.37	Sauthier *et al.* (1998)
Packed bed	Porous medium	Methanol	7.3-8.4*	Not specified	Grguric *et al.* (2000a,b)
Packed bed	Polyvinyl alcohol	Glucose	1.4	1 - 12	Park *et al.* (2000)
Packed bed	Plastic balls/ crushed oyster shells	Ethanol/methanol	6.6*	0.30 - 1.43	Menasveta *et al.* (2001)
Packed bed	Freeze-dried alginate beads	Starch	2.6	0.16	Tal *et al.* (2003a)
Digestion basin	Sludge	Endogenous	2.5	1.19	Gelfand *et al.* (2003)
Fluidized bed	Sand	Endogenous	72.6	0.015	Gelfand *et al.* (2003)
Moving bed reactor	Plastic carrier	Endogenous	15 - 24	3.5 - 5.5	Tal and Schreier (2004)
Moving bed reactor	Polyethylene	Methanol	53.0	2.5 – 3.7	Labelle *et al.* (2005)
Moving bed reactor	Polypropylene	Methanol	112.5	1.6 – 3.0	Dupla *et al.* (2006)

*extrapolated rates (rates were not provided by authors)

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CHAPTER 10

GAS TRANSFER[1]

10.0 INTRODUCTION

The availability of dissolved oxygen (DO) is usually the first factor that limits increased carrying capacity and production in intensive recirculation systems. Using only aeration as a means of providing dissolved oxygen, a system can support only about 40 kg per m^3 (0.33 lb of fish per gallon) of water. However, by using pure oxygen and high efficiency gas transfer devices to increase the amount of dissolved oxygen in the water column, stocking densities can easily be increased to over 120 kg per m^3 (1 lb of fish per gallon) of water. For example, by increasing the DO concentration at the inlet to a production tank from 10 mg/L (aeration alone) to 18 mg/L using pure oxygen, and assuming a DO concentration of 6 mg/L leaving the fish tank, the carrying capacity of the system can be increased by a factor of three. Instead of only 4 mg/L DO (10 mg/L minus 6 mg/L) being available for respiration and metabolism by the fish, 12 mg/L becomes available (18 mg/L minus 6 mg/L). Thus stocking densities can be increased from 40 kg/m^3 (0.33 lb/gal) to 120 kg/m^3 (1.0 lb/gal).

In an aquaculture system operating at these high stocking densities, with low water exchange rates, oxygenation with little aeration and low pH, often dissolved carbon dioxide, a product of fish respiration, will accumulate to levels that create toxic conditions for both the fish and the biofiltration system. Other dissolved gases that also can be important to system operation are nitrogen, argon, and hydrogen sulfide, and in some special circumstances, methane, ammonia, and radon.

In considering a treatment approach, it is important to understand that modification of the concentration of a particular gas, such as oxygen, may significantly change the concentration of other gases in solution. Thus, the design of a gas transfer system must take into consideration all the potential impacts of all the dissolved gases, as well as such water

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quality parameters as alkalinity, hardness, pH, and concentrations of iron and magnesium and of course salinity. Equations in the rest of chapter *assume freshwater* unless otherwise noted.

10.1 DISSOLVED GASES – FUNDAMENTALS

The atmosphere is composed of 20.946% oxygen (O_2), 78.084% nitrogen (N_2), 0.934% argon (Ar), 0.032%, carbon dioxide (CO_2) and other trace gases, Table 10.1. Barometric pressure (BP), or atmospheric pressure, is the sum of the partial pressures exerted by each of the individual gases in the atmosphere. The partial pressure of each gas is directly proportional to the mole fraction of that gas. Thus for oxygen in dry air at a standard temperature (20 °C) and pressure (760 mm Hg) (STP), its partial pressure is 760 mm times 0.20946 or about 160 mm Hg. Gauge pressure is equal to total pressure minus atmospheric pressure. In water, the Total Gas Pressure (TGP) is the sum of all partial gas pressures contributed by the individual gases plus the vapor pressure of water, and can be greater than or less than barometric pressure. If the ratio of the Total Gas Pressure to barometric pressure is greater than one, the water is considered to be supersaturated.

Table 10.1 Dry Air Components

Species	% volume	% mass	Molecular Wt.
Nitrogen	78.084	75.600	28.0
Oxygen	20.946	23.200	32.0
CO_2	0.032	0.048	44.0
Argon	0.934	1.300	39.9
Air	100.000	100.000	29.0

The solubility of a gas (mg/L) in water depends on its temperature, salinity, gas composition, and total pressure. The solubility of the four major gases is presented in Table 10.2 as a function of gas composition. From the Table 10.2 it can be seen that by increasing the mole fraction of oxygen from 0.20946 in air to 1.0000 with pure oxygen, the solubility or saturation concentration of oxygen is increased from 10.08 mg/L to 48.14 mg/L (in freshwater at 15°C).

Table 10.2 Solubility of Four Major Gases in Freshwater at 15 °C

Gas Species	Solubility in Air* (mg/L)	Solubility of pure gas* (mg/L)
Oxygen	10.08	48.14
Nitrogen	16.36	20.95
Argon	0.62	65.94
Carbon dioxide	0.69	1992.00

The solubility of a gas is also proportional to its absolute pressure. Increasing the gas pressure will also increase gas solubility proportionately, i.e., doubling the gas pressure will double each gas's solubility. Higher gas pressures occur in water injected into a pressurized system, or in water obtained from a deep well. One problem when using air as the oxygen source is that increasing the air pressure will also increase the solubility of nitrogen and argon above normal air saturation concentrations, which can result in potential gas supersaturation problems. Total gas pressure (TGP) for most systems should generally be kept below 105%, although the actual value is dependent on species and life stage.

Note that the gas species percentage of the makeup air will not change with elevation (up to 90 km), but the total pressure will change as noted above. In addition, assuming 100% humidity conditions will change the above numbers; water vapor pressure will be approximately 10 to 15 mm Hg and is temperature dependent.

GAS SOLUBILITY EQUATIONS

Saturation levels for dissolved oxygen, nitrogen, and carbon dioxide ($C_{s,i}$) were calculated as a function of temperature and pressure based on Henry's Law (Colt, 1984):

$$C_{s,i} = 1000 K_i \beta_i X_i \frac{P_{BP} - P_{wv}}{760} \qquad (10.1^b)$$

The Bunsen coefficients (β) and the water vapor pressure (P_{WV}) used in this determination were obtained from relationships developed by Weiss (1970, 1974) and ASHRAE (1972). The Bunsen coefficients are

[a] Symbols are defined at the end of the Chapter.

calculated separately for oxygen, nitrogen, and carbon dioxide as follows:

For Oxygen and Nitrogen in freshwater:

$$\beta_i = exp\,[-A_1 + A_2(100/T) + A_3\,ln(T/100)] \tag{10.2}$$

and for Carbon Dioxide in freshwater:

$$\beta_i = K_i\,(22.263) \tag{10.3}$$

$$K_o = exp\,[A_1 + A_2(100/T) + A_3\,ln(T/100)] \tag{10.4}$$

Temperatures (T) in Eq. 10.2 and 10.4 are in Kelvin (°C +273.15) and the A_1, A_2, and A_3 values depend on the gas.

Table 10.3 provides the mole fractions for the different gases for standard air and the necessary constants to calculate individual gas saturation values in the above equations.

Table 10.3 Constants Used in Gas Solubility Equations and Mole Fraction for Standard Air

Gas Species	Mole Fraction	K_i	A_1	A_2	A_3	J_i
Oxygen	0.20946	1.42903	58.3877	85.8079	23.8439	0.5318
Nitrogen	0.78084	1.25043	59.6274	85.7661	24.3696	0.6078
Carbon Dioxide	0.00032	1.97681	58.0931	90.5069	22.2940	0.3845

For example, assume water at 20°C, calculate the solubility of oxygen assuming a 100% oxygen environment (moist air, BP of 760 mmHg and P_{vp} of 17.54 mmHg):

$$\beta_{O_2} = \exp\left[-58.3877 + 85.8079\left(\frac{100}{293.15}\right) + 23.8439\ln\left(\frac{293.15}{100}\right)\right]$$

$$\beta_{O_2} = 0.03105\frac{L}{L\cdot atm}$$

$$C_{s,O_2} = 1000\cdot 1.42903\cdot 0.03105\cdot 1.00\frac{760-17.54}{760} = 43.35 mg/L$$

As indicated in Eq. 10.1, saturation concentration of a particular gas is proportional to the % gas composition present (in the above example, saturation is 100% or 1.00). Therefore, if the saturation concentrations for a normal atmosphere are available (as in Table 2.3, from Eq. 10.1, or from Colt, 1984), then the concentrations under pure atmospheres can be computed.

$C_{x,100\%} = C_{x\%}$ / [proportion of gas species x in the "atmosphere" in contact with the water]

Sample values for moist air, freshwater at 20 °C are:

Nitrogen: $C_{N2,\,78.08\%} = 14.88$ mg/L
$C_{N2,100\%} = 14.88$ mg/L / 0.7808 = 19.06 mg/L

Oxygen: $C_{O2\,20.95\%} = 9.08$ mg/L
$C_{O2,100\%} = 9.08$ mg/L / 0.2095 = 43.35 mg/L

Carbon Dioxide: $C_{CO2,\,0.032\%} = 0.5379$ mg/L
$C_{CO2,100\%} = 0.5379$ mg/L / 0.00032 = 1681 mg/L

As mentioned above and as shown in Eq. 10.1, saturation concentrations can also be altered by changing the Total Pressure of the gas (BP in Eq. 10.1). For example, U-Tubes increase potential dissolved oxygen levels at the bottom of the U-Tube given the increase in pressure in the water, which is proportional to the depth of the U-Tube. In summary, changes in dissolved equilibrium concentrations can be caused by manipulating pressure or gas composition.

Note: often $\frac{mg}{L}$ is expressed as parts per million, ppm

$$ppm = \frac{mg}{L} \cdot \frac{L}{1{,}000g} \cdot \frac{g}{1{,}000mg} = \frac{1}{1{,}000{,}000}$$

Note that ppm has no units or can be a ratio of the same units, e.g., lb per lb or g per g. The same units must occur in the numerator and denominator so that the ratio is "unitless". There is some small error when temperature changes the actual weight of a liter of water from 1,000 grams, i.e., liter is a volume measurement not a weight measurement.

OBSERVATIONS

Problem #1: Weather pressure front comes through and oxygen stress in fish is observed.

Explanation

Low pressure weather front caused a 20 mm Hg drop in pressure.

Atmospheric pressure is now 760 – 20 = 740 mmHg:

Applying Eq. 10.1, the new saturation concentration for oxygen (for freshwater at 20°C) is reduced to:

$$9.08\frac{mg}{L} \cdot \frac{740 - 17.54}{760 - 17.54} \approx 9.08 \cdot \frac{740}{760} = 8.84\frac{mg}{L}$$

Problem #2: Insufficient oxygen available in tank system. Gas conditioning device used to add oxygen is using pure air. Grower needs to double oxygen being added.

Solution

Increase the gaseous concentration of oxygen in the "device" to <u>2 times</u> atmospheric or approximately 42%. All other things remaining approximately constant, this could "double" oxygen being transferred into the fish tanks. The actual quantity of oxygen transferred into the fish water will depend upon several factors, including the concentration of the oxygen entering the gas conditioning device.

BAROMETRIC PRESSURE (P_{BP})

An estimate of barometric pressure as a function of elevation can be obtained using the following equation (Colt, 1984)

$$P_{BP} = 10^{a} \qquad \text{where} \quad a = 2.880814 - \frac{h}{19{,}748.2} \tag{10.5}$$

h is the height above sea level in meters

For example, for an elevation of 300 m above sea level, the atmospheric pressure is calculated as follows using Eq. 10.5:

$$a = 2.880814 - \frac{300}{19{,}748.2} = 2.865562$$

$$P_{BP} = 10^{2.865562} = 734\ mm\ Hg$$

WATER VAPOR PRESSURE (P_{WV})

Data given by Colt (1984) was used to regress water vapor pressure versus temperature between 10 and 30°C ($R^2 = 0.999$)

$$P_{WV} = A_0 e^{0.0645T} \tag{10.6}$$

where A_0 is 4.7603for temperature in °C.

For example, the vapor pressures for water at 0, 10 and 30°C are 4.7, 9.1 and 33.0 mm Hg, respectively, using Eq. 10.6 (see Table 2.1 or A-14).

SUPER SATURATION OF GASES

The partial pressure of a gas (P_i^1, mm Hg) given a measured concentration level in the liquid phase ($C_{meas,i}$, mg/L) can be calculated as follows (mm Hg) (Colt, 1984):

$$P_i^l = \left[\frac{C_{meas,i}}{\beta_i}\right] J_i \qquad (10.7)$$

where values of J_i are given also in Table 10.3 and β_i is the Bunsen coefficient, calculated based upon the constants given in the same table.

Partial pressure of a gas in the gas phase (mm Hg) is proportional to mole fraction of gas:

$$P_i^g = X_i(P_{BP} - P_{WV}) \qquad (10.8)$$

Users often prefer to view dissolved gas concentrations as its percent of saturation:

$$S_{\%} = \left[\frac{P^l}{P^g}\right] 100 \qquad (10.9)$$

Saturation concentration as a percentage is also equal to the ratio between the measured concentration and saturation concentration, both in mg/L:

$$S_{\%} = \left[\frac{C_{meas,i}}{C_{s,i}}\right] 100 \qquad (10.10)$$

TOTAL GAS PRESSURE[b]

Finally, of particular importance to fish culture is the total gas pressure (P_{TG}) of the combined gases in solution:

$$P_{TG} = P^l_{oxygen} + P^l_{nitrogen} + P^l_{CO_2} + P_{wv} \qquad (10.11)$$

The partial pressure of Argon is included explicitly in some cases. We often have fish culture recommendations based around an acceptable P_{TG} expressed as a percentage of barometric pressure:

$$P_{TG}(\%) = \frac{P_{TG}}{P_{BP}} \cdot 100 \qquad (10.12)$$

[b] total gas pressure is often noted as TGP

EXAMPLE: USE OF GAS EQUATIONS

The equations previously presented are a bit tedious to say the least. For sake of demonstration and to provide numbers for others to compare to once adapting these equations to spread sheet or computer type formats, some sample calculations are provided.

Assume: freshwater at 25°C and 228.6 m elevation

Barometric and Water Vapor Pressures

$P_{BP} = 10^{(2.880814 - h/19748.2)}$
$P_{BP} = 10^{(2.880814 - 228.6/19748.2)}$
$P_{BP} = 740$ mm Hg

$P_{wv} = 4.7603\ e^{(0.0645)(25)}$
$P_{wv} = 23.87$ mm Hg

Oxygen

$K = 1.42903$

$\beta = \exp[A_1 + A_2(100/T) + A_3\ln(T/100)]$
$\beta = \exp[-58.3877 + 85.8079(100/298.15) + 23.8439\ln(298.15/100)]$
$\beta = 0.02844$ L/L-atm

$X = 0.20946$
$C = 1000KBX(P_{BP} - P_{wv})/760$
$C = 1000\ (1.42903)\ (0.02844)\ (0.20946)\ (740 - 23.87)/760$
$C = 8.022$ mg/L

Nitrogen

$K = 1.25043$

$\beta = \exp[A_1 + A_2(100/T) + A_3\ln(T/100)]$
$\beta = \exp[-59.6274 + 85.7661(100/298.15) + 24.3696\ln(298.15/100)]$
$\beta = 0.01442$ L/L-atm

$X = 0.78084$

$C = 1000KBX(P_{BP} - P_{wv})/760$
$C = 1000\,(1.25043)\,(0.01442)\,(0.78084)\,(740 - 23.87)/760$
$C = 13.264$ mg/L

Carbon Dioxide

$K = 1.97681$

$\beta = K_o\,(22.263)$
$K_o = \exp[-58.0931 + 90.5069(100/298.15) + 22.2940\ln(298.15/100)]$
$K_o = 0.03397$

$\beta = (0.03397)\,(22.263)$
$\beta = 0.7562$ L/L-atm

$X = 0.00032$ (or 320 ppm)
$C = 1000KBX(P_{BP} - P_{wv})/760$
$C = 1000\,(1.97681)\,(0.7563)\,(0.00032)\,(740 - 23.87)/760$
$C = 0.451$ mg/L

Now assume that we measured the following dissolved gas conditions for our site (25°C and 228.6 m elevation):

$C_{oxygen} = 6.0$ mg/L
$C_{nitrogen} = 13.3$ mg/L
$C_{carbon\ dioxide} = 80.0$ mg/L

We can calculate individual partial pressures corresponding to the measured gas concentrations in the liquid and then compare them to their saturation values:

$$P^l = \left[\frac{C_{meas}}{\beta}\right] J_i$$

Oxygen

$$P^l = \left[\frac{6.0}{0.02844}\right]0.5318$$

$P^l = 112.2$ mm Hg

$P^g = X(P_{BP} - P_{wv})$
$P^g = 0.20946(740 - 23.87)$
$P^g = 150.0$ mm Hg

$S_{\%} = (P^l/P^g)100$
$S_{\%} = (112.2/150)100$
$S_{\%} = 74.8\%$

Nitrogen

$$P^l = \left[\frac{13.3}{0.01442}\right]0.6078$$

$P^l = 560.6$ mm Hg

$P^g = X(P_{BP} - P_{wv})$
$P^g = 0.78084(740 - 23.87)$
$P^g = 559.2$ mm Hg

$S_{\%} = (P^l/P^g)100$
$S_{\%} = (560.6/559.3)100$
$S_{\%} = 100.2\%$

Carbon Dioxide

$$P^l = \left[\frac{80.0}{0.7563}\right]0.3845$$

$P^l = 40.67$ mm Hg

$P^g = X(P_{BP} - P_{wv})$
$P^g = 0.00032(740 - 23.87)$
$P^g = 0.23$ mm Hg
$S_{\%} = (P^l/P^g)100$
$S_{\%} = (40.67/0.23)100$
$S_{\%} = 17{,}682\%$

$P_{TG} = P^{l}_{oxygen} + P^{l}_{nitrogen} + P^{l}_{carbondioxide} + P_{wv}$
$P_{TG} = 112.2 + 560.6 + 40.67 + 23.87$
$P_{TG} = 737.34$ mm Hg

$P_{TG}\ (\%) = (737.23/740)100$
$P_{TG}\ (\%) = 99.6\%$

Note that in the above example, argon has been omitted and would have contributed 6.82 mm Hg to the total gas pressure if it had been at saturation concentration. This corresponds to an increase in the P_{TG} to (737.34 + 6.82) = 744.16 mm Hg, or $P_{TG}(\%)$ = 100.6%, making the P_{TG} > atmospheric pressure. The reader can refer to Colt (1984) if these additional equations are required for their particular applications.

10.2 GAS TRANSFER

TRANSFER OF GASES

Gas Transfer is proportional to pressure difference for each particular gas. A species gas will move into a "higher" total pressure environment if the partial pressure of the species gas is higher than the species gas pressure in the "other" environment.

Principle: Gas transfer occurs due to *pressure differences*.

$$Q_i \frac{mass}{time} = \frac{1}{R_{pressure}} \cdot (P_{i,high} - P_{i,low}) \tag{10.13}$$

The rate of gas flow from a high pressure area to a lower pressure area is determined by the *resistance* to gas flow from the high to low pressure areas.

VALUES FOR PRESSURE

There are several ways to give atmospheric pressure, mostly depending upon your own country's preference and the unit system you are most comfortable with. The most common terms used to describe atmospheric pressure are:

At sea level 760 mm (29.91 inches) of mercury (Hg)
34 feet of water column
14.96 psi

TERMS USED TO DESCRIBE PRESSURE

Atmospheric Pressure is a sum of the ***partial pressures*** of contributing gases and the vapor pressure of water. *Total gas pressure (TGP)* is also the sum of all the partial pressures (there may be non-typical gas concentrations for an individual gas component) and the vapor pressure of water (Eq. 10.10). Total Gas Pressure should not exceed 105% of atmospheric pressure (species dependent). Pure oxygen systems and green water systems can be subject to excessive total gas pressure problems. Incomplete degasification of "incoming" water, particularly for nitrogen, can lead to excessive total gas pressure (causes gas bubble disease in fish), since incoming water sources could be and often are supersaturated with nitrogen gas.

$$\text{Gauge Pressure} = \text{Total Pressure} - \text{Atmospheric Pressure} \tag{10.14}$$

FUNDAMENTALS OF GAS TRANSFER

When air is in contact with water, dissolved gases in the water attempt to reach equilibrium with the partial pressures of the gases in the atmosphere. Two factors that directly impact the rate of gas transfer are first the area of gas-liquid interface and second the difference between the concentration (partial pressure) at saturation and the existing concentration of the gas in the water. For example, if the water is under saturated with the gas in question (a deficit), the gas will be transferred into solution and if the water is supersaturated, out of solution. In a simple trickling tower, it is possible to have supersaturated nitrogen gas being removed, while under saturated dissolved oxygen, increases in concentration. This overall gas transfer rate is dependent on the deficit (or surplus) of a dissolved gas and a proportionality constant, usually called the gas transfer coefficient. The overall gas transfer coefficient represents conditions in a specific gas transfer system. It is a composite term that includes such factors as the diffusion coefficient for the gas, the liquid-film thickness, and the area of gas-liquid interface. These factors also suggest ways to increase the overall rate of gas transfer. This could be accomplished, for example, by decreasing the liquid-film thickness by turbulence or mixing, increasing the gas-liquid interface by making the bubble size smaller, or increasing the concentration gradient.

In order to characterize the gas transfer, or more specifically the oxygen transfer ability of different system designs, a set of standard conditions and a standardized tests regime has been developed. The standard oxygen transfer rate (SOTR), defined as the amount of oxygen (kg O_2/hr) that a device will transfer to clean water at 0 mg/L DO and

20°C, is determined from these tests. (SOTR can be converted to pounds of oxygen per hour by multiplying by 2.205.) In addition, the amount of oxygen transferred by a device per unit of energy consumption (kg O_2/kWh) is defined as the standard aeration efficiency (SAE). Finally, the standard oxygen transfer efficiency (SOTE) or the absorption efficiency (AE) is defined as the oxygen transferred divided by the mass flow rate of oxygen supplied to the gas transfer device. Table 10.4 provides the correction factors for SOTR when conditions depart from the standard initial conditions of 0 mg/L DO and 20°C.

Table 10.4 SOTR[a] Correction Factors for Other Water Temperatures or Initial DO Levels: OTR = Correction Factor x SOTR; (Note table data was developed from experimental data using pond waters and extracted from a nomograph; thus there is an unexpected lower value for the correction factor at 70F and 0 mg/L initial condition).

Dissolved Oxygen in Tank Water (ppm)	Tank Water Temperature in °F				
	50	60	70	80	90
0	.90	.91	.92	.93	.98
1	.82	.82	.82	.81	.95
2	.74	.73	.71	.69	.71
3	.66	.63	.61	.57	.57
4	.58	.54	.50	.45	.43
5	.49	.44	.39	.33	.29
6	.41	.35	.29	.21	.15
7	.33	.26	.18	.08	.02
8	.25	.16	.07	.00	.00
9	.17	.07	.00		
10	.09	.00			
11	.00				

[a] Ratings Obtained at Standard Conditions of 0 mg/L and 68° F (LSU, 1988)

The corrections factors shown in Table 10.4 are somewhat non-intuitive, since the table shows a correction factor of 0.90 for an initial zero value of oxygen and at a temperature of 70F (close to standard conditions $C_{s,std}$ of 68 F and 0 mg/L oxygen). The values then also increase as temperature increases to near unity (0.98 at 90F). This is probably due to the α factor in (Eq. 10.16) previously mentioned.

An alternative approach to calculate the correction factor for non-standard operating conditions, where C_s is saturation concentration and T in Celsius, is:

$$\text{Correction Factor} = \left[\frac{C_{s,T} - C}{C_{s,std} - 0}\right] * 1.024^{(T-20)}$$

Overall gas mass transfer in a variety of devices can be represented by a G value as described originally by Colt and Bouck (1984), in an equation developed to predict gas transfer in packed columns:

$$G_{20} = \ln\left[\frac{C_s - C_{in}}{C_s - C_{out}}\right] \tag{10.15}$$

The G value is very useful in evaluating the relative effectiveness of any particular gas transfer device. Once the G value is established for the device, it can then be used to predict gas transfer for any particular set of initial conditions of C_{in} and C_s. The effects of temperature (°C) can be adjusted for using a van't Hoff-Arrhenius relationship (APHA, 1995):

$$G_T = G_{20}\,\alpha\,(1.024)^{T-20} \tag{10.16}$$

Knowing influent conditions, Eqs. 10.14–16 can be used to calculate effluent conditions since effluent concentration is the only unknown in the equation:

$$C_{out} = C_s + (C_{in} - C_s)\,e^{-G_T} \tag{10.17}$$

The α factor in (10.16) represents the increase in gas-liquid interfacial resistance to diffusion due to surface active compounds (Stenstrom and Gilbert, 1981). The α value is considered to be unity for clean water, but has been reported as 0.92 for surface waters from a reservoir (Ahmad and Boyd, 1988). Adjustments for G values for nitrogen and carbon dioxide are based upon the molecular diameters of the gas species relative to oxygen. Tsivoglou et al. (1965) applied Einstein's law of diffusion to estimate that gas transfer for different gases is inversely proportional to the ratios of the molecular diameters. Applying this theory means that nitrogen gas transfer is 94% and carbon dioxide transfer is 90% of the rates occurring for oxygen.

Gas transfer efficiency values, E, are calculated based upon the change in DO across the system, expressed as a percentage of the initial dissolved gas deficit (Downing and Truesdale, 1955):

$$E = \left[\frac{DO_{out} - DO_{in}}{C_s - DO_{in}} \right] \cdot 100 \tag{10.18}$$

Traditionally, gas transfer efficiency has been used to describe the effectiveness of various apparatus (see Table 10.5). Equation 10.19 relates E and G.

$$ln \left[\frac{DO_{out} - DO_{in}}{C_s - DO_{out}} \right] \cdot \frac{1}{E} = G \tag{10.19}$$

Table 10.5 Aeration Effectiveness of Various Devices

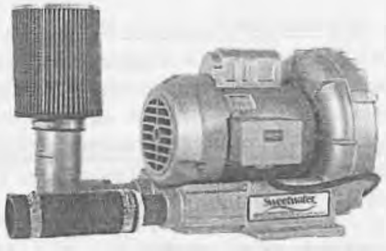
Gas Transfer Device	Fall Distance (cm)	Aeration Efficiency (E,%),
Simple Weir	23	6.2
	30	9.3
	67	12.4
Inclined Corrugated Sheet	30	25
	61	43
Splash Board	23	14
	30	24
	67	38
Cascade	25	23
	50	33
	100	52
Single Orifice	na	10
Packed Columns		see Eq. 10.20
Spray Towers		see Eq. 10.21

Oxygen absorption efficiency is calculated as the ratio of oxygen absorbed in a particular device to the amount of oxygen added and expressed as a percentage.

10.3 Gas Transfer Options

Aeration Systems – Air Stones, Packed Towers

Sources of Air



The standard sources of air in aquaculture are blowers, air pumps, or compressors. The primary differences between them are the pressure requirements and the volume of the discharge. Blowers supply high volumes of air at low pressure, while compressors supply small volumes of high pressure air. In specifying the type of air source required, two design parameters need to be determined: the required pressure and the required air volume. The operating pressure is determined by the requirements to overcome the water pressure at the diffuser's depth, the pipe friction losses, and the diffuser's resistance to air flows. For a typical application of air stones in a shallow (~ 1 m deep) tank, this is about 125 mm Hg (2 to 3 psi). In deeper tanks or with diffusers requiring higher pressures, i.e., those with smaller bubbles or clogged pores, this could be considerably higher. The air volume required is determined by the pounds of oxygen required and the overall transfer efficiency of the system. For example, a 23 cm (9 inch) air stone operating in 1 m (3 feet) of water with 1.2 m^3/hr (0.7 cfm) air supply transfers only 0.25 kg/day (0.023 lbs/hr) of oxygen.

Regenerative blowers are designed to provide large volumes of air at low pressure, typically less than 190 mm Hg (4 psi). They are most commonly used with either air stones or airlift systems. Advantages of regenerative blowers include their low noise levels, reliability, energy efficient motors, and lower comparative cost. Air pumps operate in the mid-range of performance, between blowers and compressors. Compressors are designed for high pressure operations, such as in very deep tanks or where long airlines are required.

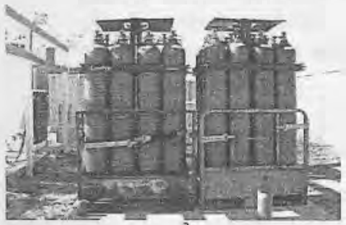
Air Stones

Air stones are very inefficient oxygen transfer devices (3–7%), but very inexpensive in terms of capital and operating costs. At low stocking

densities and high exchange rates, they work very well at maintaining adequate oxygen levels. One disadvantage is the maintenance requirements due to clogging and biofouling, especially in very hard water.

OXYGEN TRANSFER SYSTEMS

SOURCES OF OXYGEN



In aquaculture, three sources of oxygen are commonly used: high-pressure oxygen gas, liquid oxygen (LOX), and on-site oxygen generations. To insure availability and as backup, usually at least two sources are available at most facilities. High pressure oxygen gas is easily available in cylinders containing from 3 to 7 m^3 (100 to 250 ft^3) of gas at 170 atmospheres of pressure (2550 psi). A number of cylinders can be connected together using commercially available manifolds to increase the total capacity. Due to their cost and limited capacity, oxygen cylinders are normally used only as emergency backup systems.

In many areas, liquid oxygen is commercially available in bulk and can readily be transported and stored in on-site Dewar's type storage containers. At one atmosphere, liquid oxygen boils at -182.96°C (-297.3°F), thus special insulated cryogenic containers are required for storage. These containers range in size from 0.11 m^3 liquid (30 gal) to a much as 38 m^3 liquid (10,000 gal), and are usually rented or leased from the suppliers, although the smaller units can be purchased. One gallon of liquid oxygen is equal to 3.26 m^3 (115 ft^3) of gaseous oxygen. The maximum gas pressure in these containers is in the range of 10 to 14 atmospheres (150 to 200 psi). Prior to its use, the LOX is vaporized by directing it through heat exchanger coils. A liquid oxygen supply system will consist of a storage tank, vaporizer, filters, and pressure regulators. The economics of LOX use are dependent upon the transport cost, and the reduced capital and maintenance cost as compared to pressure swing adsorption

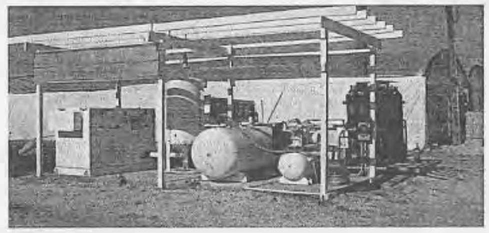
(PSA) or vacuum swing adsorption (VSA) systems. In general, a LOX system is very reliable, operating even during power failures. Failures on farms using LOX systems as backup to power outage are caused by under-sizing the LOX system in the first place or unanticipated severe weather conditions that extend longer than predicted. Carefully consider your risks for such cases and size your LOX system with these potential dangers in mind. As a minimum, a LOX system should be able to maintain a facility with oxygen for 30 days. Remember that upon the first sign of major weather problems, it is probably prudent to take your fish off of feed, which will lower their oxygen demand dramatically over the next 24 hours.

"Rule of Thumb"
Oxygen Parameters

1 lb liquid oxygen = 342 liters (gas)
1 lb liquid oxygen = 12.1 ft^3 (gas)
1 gallon liquid oxygen = 115 ft^3 (gas)
1 L liquid oxygen = 0.86 m^3 (gas)

Gas Phase:
Volume @ 70° F and 1 atm: 0.7513 m^3/kg (12.08 ft^3/lb)
Density @ 70° F and 1 atm: 1.33 kg/m^3 (0.0828 lbs/ft^3)

Liquid Phase:
Specific Volume: 0.877 L/kg (0.105 gal/lb)
Density: 1.141 kg/L (9.52 lbs/gal)



Oxygen can also be generated on-site using either a pressure swing adsorption (PSA) or a vacuum swing adsorption (VSA) unit. In both cases, a molecular sieve material is used to selectively adsorb or absorb nitrogen from the air, producing an oxygen-enriched gas. Commercially available units can produce anywhere from 0.5 to 14 kg (1 to 30 lbs) of oxygen per hour at from 0.7 to 3.3 atmospheres (10 to 50 psi). A source of dry, filtered air at 6.0 to 10.0 atmospheres (90 to 150 psi) is required to produce an oxygen stream that is from 85–95% pure.

PSA and VSA units operate on a demand basis and produce oxygen only when needed. They have proven to be very reliable and require little maintenance. However, they can be expensive in terms of capital and operating expenses, due to the compressed air requirements. Such units appear to be most competitive for smaller operations, e.g., production of less than 100 tons per year. Also, since they require electrical power, some other source of oxygen is needed in the event of power failures or else the facility must be equipped with large backup generators and transfer switches. Properly designed gas storage systems can also be implemented with some of these systems.

U-TUBES

The U-tube aerator operates by increasing the gas pressure, thus increasing the overall gas transfer rate. It consists of either two concentric pipes or two pipes in a vertical shaft 9 to 45 m (30 to 150 ft) deep. Oxygen is added at the upper end of the down-leg of the U-tube and as the water/gas moves downward through the contact loop, an increase in hydrostatic pressure increase the oxygen transfer rate. The overall oxygen transfer efficiency is a function of the depth of the U-tube, inlet gas flow rate, water velocity, diffuser depth, and inlet DO concentration. Concentrations of dissolved oxygen ranging from 20-40 mg/L can be achieved, but the overall oxygen transfer efficiency if only 30–50%. Off-gas recycling can improve the absorption efficiency to 55–80%. Two advantages of the U-tube are the low hydraulic head requirements that allow operation with no external power if sufficient head is available, and that it can be used with water containing high levels of particulates or organics. Its chief disadvantages are that it does not vent off gasses such as nitrogen or carbon dioxide very efficiently and construction costs can be high, particularly if bedrock is present.

"Rule of Thumb"
U-Tube Aerator
Design U-Tubes for a down flow velocity between 2 m/s to 3 m/s
Limit G/L ratio to <25%

U-tubes are designed for flows where the downflow velocity is between 1.8 to 3.0 m/s. A particularly unique problem with U-tubes is that if too much oxygen is added a gas bubble blockage can occur that results in flow interruption. This will tend to happen if gas-liquid ratios exceed 25%. Be careful when adding oxygen. A U-tube should never be used with air as the injected gas due to the risk of total gas supersaturation.

For example, a U-Tube that was 12 m (40 feet) in depth would increased the potential for absorbing oxygen as follows (atmospheric pressure is equivalent to 10.4 m (34 feet of water) for freshwater at 15°C:

$$C_{oxygen,12.2m} = C_{oxygen,surface} \cdot \left(\frac{P_{atm} + P_{hydrostatic}}{P_{atm}} \right)$$

If the gas is air:

$$C_{oxygen,12.2m} = 10.17 \frac{mg}{L} \cdot \left(\frac{10.4m + 12.2m}{10.4m} \right) = 22.1 \frac{mg}{L}$$

If the gas is oxygen (100% oxygen vs. 21% oxygen in air):

$$C_{oxygen,12.2m} = 10.17 \frac{mg}{L} \cdot \frac{1.00}{0.21} \left(\frac{10.4m + 12.2m}{10.4m} \right) = 105.2 \frac{mg}{L}$$

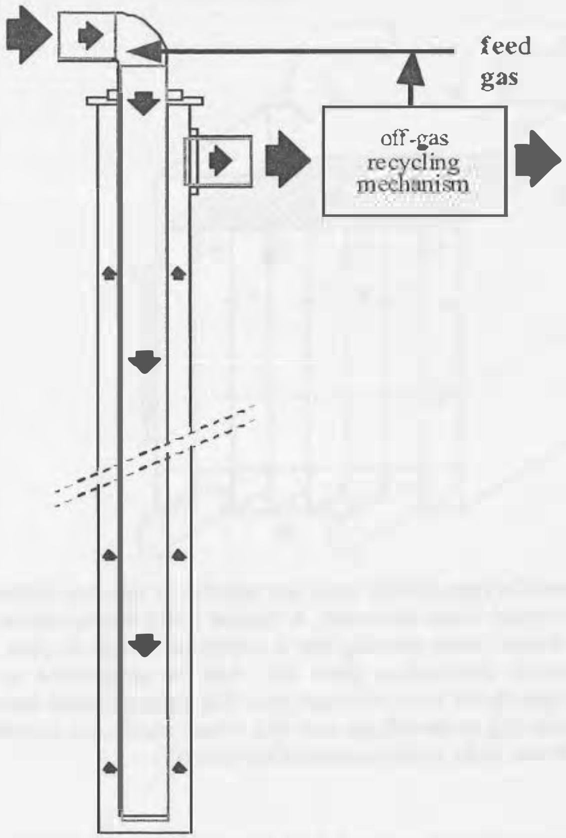


Figure 10.1 U-Tube aeration system.

PACKED COLUMNS

Packed columns consist of a vertical column filled with media having a high specific surface area. Water is uniformly distributed over the top of the media with a perforated plate or through a spray bar, and trickles down through the media. Oxygen is injected into the column and is transferred into the passing water through the large gas/liquid interface on the media. The column may be either open or closed at the top. Packed columns are efficient nitrogen and carbon dioxide strippers if air is used as the injected gas rather than oxygen. However, a high gas/liquid ratio (forced aeration) is needed for carbon dioxide stripping. Packed columns are simple to build and easy to retrofit into existing facilities.

Performance design characteristics include the water distribution method, media characteristics, media bed depth, gas/liquid loading rates, inlet DO concentration and operating pressure. Their main disadvantage is fouling due to the accumulation of organics and particulates on the media over time.

Packed columns have two additional advantages:

- Provide nitrification
- Provide CO_2 gas stripping

The above features allow to some degree that a packed tower can serve as the complete water conditioning system for lightly loaded systems. CO_2 stripping is discussed in more detail later in this Chapter.

Hydraulic loading rates should be at least 7 L/m²s (10 gpm/ft²), and can be as high as 48 L/m²s (70 gpm/ft²). Excessive hydraulic loadings will cause the packing to be flooded and must be avoided.

The gas transfer efficiency for packed columns depends upon the tower packing height (Z), the hydraulic loading (HL), and the characteristics of the packing material. Performance of packed columns can be predicted based upon the data developed by Watten (1990) and given in Table 10.6. The general form of the prediction equation is:

$$G_{20} = a + b \cdot Z \qquad (10.20)$$

where the coefficients in Eq. 10.20 are given in Table 10.6

Closely related to a packed column and included within this section are spray towers. These units are also some type of tower or column but they are enclosed and then some type of spray nozzle is placed within them to provide the water gas breakup and gas transfer. Vinci et al. (1997) developed the following predictive equation using a 20 cm column diameter and a full cone spray nozzle (Spraying Systems Co., Wheaton, IL):

$$G = -0.05 + 0.3025 \cdot Z_{tower} + 0.000067\,HL \qquad (10.21)$$

Table 10.6 Coefficients for Predicting Packed Column Performance as Affected by Hydraulic Loading (HL, reported in L/m^2s) (Watten, 1990)

Packing Type	Geometric Surface Area, m^2/m^3	Coefficient a	Coefficient b
2.54 cm TriPack®	279		
HL[a]=32		0.327	1.655
HL=61		0.277	1.589
3.81 cm Nor-Pac®	144		
HL=32		0.324	1.555
HL=61		0.398	1.428
3.81 cm ACTIFIL®	139		
HL=34-73		0.357	1.349
5.08 cm Nor-Pac®	102		
HL=32		0.243	1.285
HL=61		0.162	1.855

[a]HL Coefficient a & b refer to Equation 10.20

LHO's

Low Head Oxygenators (LHO) are being used more frequently, particularly because of their adaptability to high flows using minimal hydraulic head, hence their name Low Head Oxygenator. The original LHO design was developed and patented by Watten (1989). LHO's vary in configuration, but all are fundamentally similar in operation. These units consist of a distribution plate positioned over multiple (5 to 10) rectangular chambers, Fig. 10.2. Water flows over the dam boards at the end of a raceway or is pumped upwards from an indoor fish tank, through the distribution plate, and then falls through the rectangular chambers. These chambers provide the gas-liquid interface needed for mixing and gas transfer. The streams of falling water impact a collection pool at the bottom of each chamber where the effluent water flows away from each chamber equally in parallel. All of the pure oxygen is introduced into the outer or first rectangular chamber. The mixture of gases in the first chamber, which now has a diluted oxygen concentration passes, sequentially through the remaining chambers. The gaseous mixture will decrease in oxygen concentration from chamber to chamber as the oxygen is continued to be absorbed. Finally the gaseous mixture will exit from the last chamber. This gas is referred to as off-gas. Each of the rectangular chambers is gas tight and the orifices between the chambers are properly sized and located to reduce back-mixing between chambers.

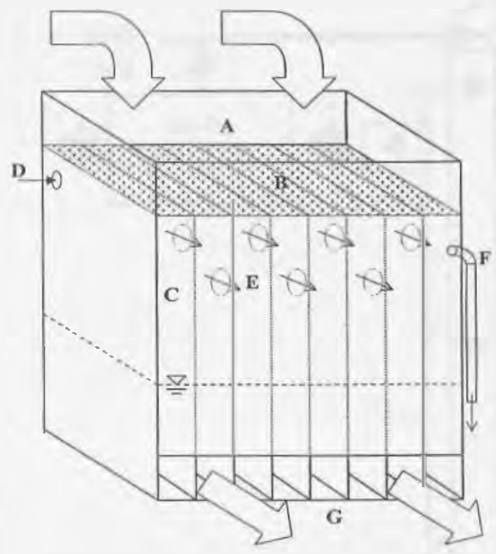


Figure 10.2 Low Head Oxygen (LHO) units are popular in raceway culture to restore oxygen in serial reuse raceways. A typical LHO configuration and components are shown: water flowing into a collection trough or plate (A), through a perforated distribution plate (B), and is oxygenated in the chambers (C), as gas flows from inlet gas port (D), through holes between chamber to chamber (E), to the off gas port (F), where excess gas is bubbled off under water. Water exits at the bottom of the unit (G).

The operating performance of an LHO is primarily affected by the hydraulic head on the distribution plate (Y_1), the orifice hole size (Y_2), the depth of the receiving pool (Y_3), and the drop or fall distance from the orifice plate to the receiving pool of water (Y_4). Davenport et al. (2001) developed a regression model to predict the overall gas transfer coefficient G_{20} as a function of these geometric variables for a single chamber:

$$G_{20} = -0.0059(Y_2) + 0.017(Y_3) + 0.011(Y_4) - 0.00047(Y_3^2) - 0.000034(Y_4^2) + 0.00034(Y_2\,Y_3) - 0.000049(Y_2\,Y_4) + 0.000026(Y_3\,Y_4) \quad (10.22)$$

If (Y_3 >41 cm) Then $Y_3 = 41$
If (Y_2 >19 cm) Then $Y_2 = 19$

where hole size is in mm diameter and pool depth and drop height are in cm.

Timmons et al. (2001) developed an LHO model that is provided in the book's software package and is described further in the appendix. The Timmons LHO model calculates LHO performance as affected by hole size, hydraulic head over the flooded plate, hole orifice discharge coefficient (C_d), percent active hole area of the plate, plate size, and the number of chambers involved:

$$Q = C_d A\sqrt{2gY_1} \tag{10.23}$$

The value for C_d is predicted using the following equation (Timmons et al. 2001)

$$\begin{aligned} C_d = {} & 0.914 - 0.00308Y_2 - 0.0519Y_1 + 0.000228Y_2^2 \\ & + 0.00298Y_1^2 - 0.000660Y_2Y_1 \end{aligned} \tag{10.24}$$

If(Y_1 >13) Then Y_1 = 13
If(Y_1 <2.5) Then Y_1 = 2.5

A graphical representation of Eq. 10.24 is shown in Fig. 10.3. Classical approaches can be used to calculate C_d as well (Streeter, 1966) based upon jet Reynolds number and ratio of upstream cross sectional area to hole area.

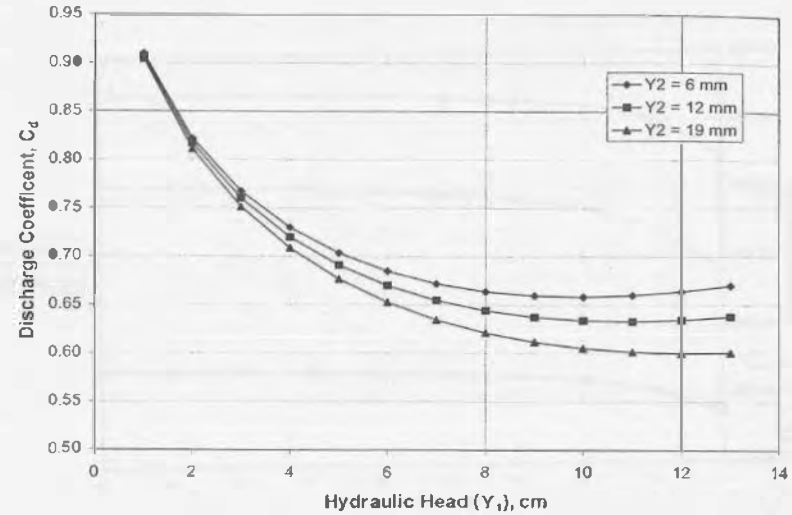


Figure 10.3 Discharge coefficient related to hole diameter (Y_2) and hydraulic head (Y_1).

Hole Size

The proper size of the holes to be used in the distribution plate can be determined by knowing the type of system the LHO will serve. Smaller holes generally have greater potential efficiencies. Smaller holes are also an advantage because the momentum of the water jets impacting the collection pool is less and the resulting gas bubbles that are formed will not travel downwards as far. This is advantageous because bubbles that become entrained in the effluent flow and do not rise back into the LHO chamber will be wasted to the atmosphere increasing operational cost. The trade off is that smaller holes will more readily clog from biological fouling or particulates. Some consideration needs to be given at this stage as to the nature of the system the LHO is serving. Potential sources of clogging particulates will be fish feces and scales. In general, the holes should be sized as small as possible but not so small as to incur frequent plugging.

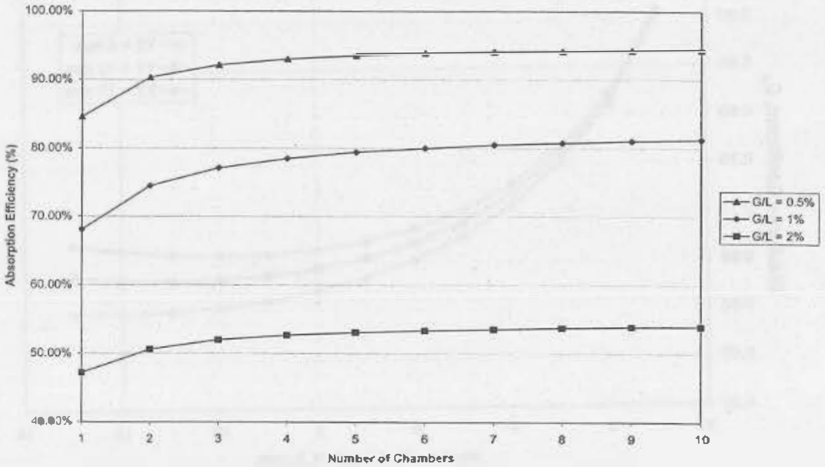


Figure 10.4 Absorption efficiency as affected by the number of LHO chambers and the gas liquid G/L ratio (Arbitrary Set of model inputs were: Y_2 = 9.5 mm; Y_3 = 13 cm; Y_4 = 61 cm; Y_1 = 7.5 cm; Temp = 20.0°C; Top Area = 0.1 m^2; Active Hole Area = 10.0%; Chambers = Varies; G/L = Varies; DO_{in} = 6.0 mg/L; DN_{in} = 14.0 mg/L; DCO_2 = 0.0; Pressure = 760.0 mm Hg; Oxygen Fraction in Inlet Gas = 0.99).

Predictions of LHO Performance

The LHO model is used to demonstrate the effects of LHO chamber number on gas absorption efficiency and effluent DO for various G/L ratios, Figs. 10.4 and 10.5, for an arbitrary LHO configuration (Y_1 = 7.5 cm; Y_2 = 9.5 mm; Y_3 = 13 cm; Y_4 = 61 cm; Temp = 20.0 °C; Top Area = 0.1 m^2; Active Hole Area = 10.0%; DO_{in} = 6.0 mg/L; DN_{in} = 14.0 mg/L; $DCO_{2,\,in}$ = 0.0; Atmospheric Pressure = 760.0 mm Hg; Oxygen Fraction in Inlet Gas = 0.99). As can be seen in these two figures, an LHO should have at least 4 or 5 chambers to obtain high gas transfer efficiency. This is reflected in current commercial units that typically have 7 chambers. It is also quite evident from Fig. 10.4 that gas transfer efficiency is severely degraded at a G/L ratio of 2% (slightly over 50%). Thus, increasing G/L ratios to obtain higher effluent DO to meet biological fish demand is not an economical choice. In fact, the producer would probably be economically ahead to reduce fish density rather than to try to maintain the higher densities by using elevated G/L flow rates.

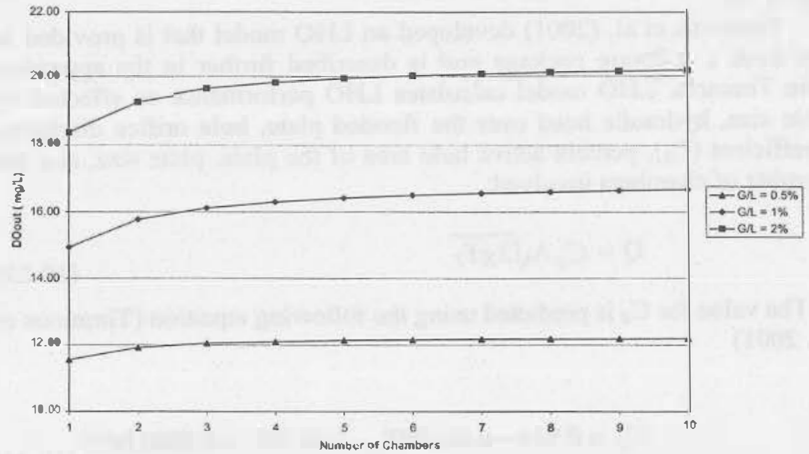


Figure 10.5 Effluent DO as affected by the number of LHO chambers and the gas liquid G/L ratio (Arbitrary set of model inputs were: Y_2 = 9.5 mm; Y_3 = 13 cm; Y_4 = 61 cm; Y_1 = 7.5 cm; Temp = 20.0°C; Top Area = 0.1 m^2; Active Hole Area = 10.0%; Chambers = Varies; G/L = Varies; DO_{in} = 6.0 mg/L; DN_{in} = 14.0 mg/L; DCO_2 = 0.0; Pressure = 760.0 mm Hg; Oxygen Fraction in Inlet Gas = 0.99).

The effect of G/L ratio on absorption and effluent DO is demonstrated explicitly in Fig. 10.6 for the standard LHO unit selected as noted above. This graph indicates that a 1.4% G/L ratio is the largest gas flow that could be used if one were trying to achieve a minimum oxygen absorption efficiency of 70%; this would correspond to an increase in the effluent DO by 12 mg/L over the influent DO value of 6 mg/L. A rule of thumb that emerges from this is that delta DO's of 10 to 12 are target values for operating LHO units. The rapid drop in absorption efficiency as G/L ratios are increased is also a clear warning to the aquaculturalist that LHO gas usage should be closely monitored to avoid the easy (but expensive!) solution of simply increasing G/L to increase effluent DO. The sensitivity of effluent DO and gas transfer efficiency to G_{20} is demonstrated in Fig. 10.7. This graph was created by assigning G_{20} values to the computer model instead of calculating them using Eq. 10.21 See the appendix for sample output screens and user instructions for employing the LHO model.

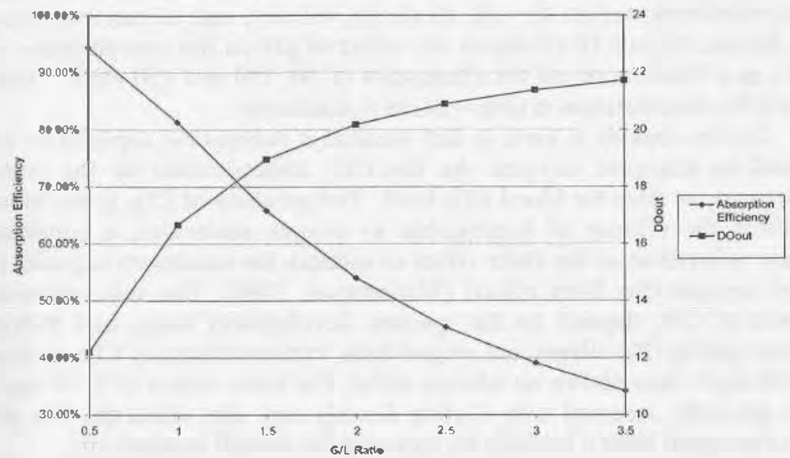


Figure 10.6 Absorption efficiency and effluent DO as affected by gas-liquid ratio G/L (Arbitrary set of model inputs were: Y_2 = 9.5 mm; Y_3 = 13 cm; Y_4 = 61 cm; Y_1 = 7.5 cm; Temp = 20.0°C; Top Area = 0.1 m^2; Active Hole Area = 10.0%; Chambers = 10; G/L Varies; DO_{in} = 6.0 mg/L; DN_{in} = 14.0 mg/L; DCO_2= 0.0; Pressure = 760.0 mm Hg; Oxygen fraction in inlet gas = 0.99).

AERATION CONE OR DOWN-FLOW BUBBLE CONTACTORS

The aeration cone, Speece cone, or downflow bubble-contact aerator consists of a cone-shaped cylinder or a series of pipes with reducing diameters. Water and oxygen enter at the top of the cone, flow downward, and out. As the cone's diameter increases, the water velocity decreases, until the downward velocity of the water equals the upward buoyant velocity of the bubbles. Thus, the bubbles are held in suspension, until they dissolve into the water. The performance of aeration cones is determined by gas and water flow rates, influent DO concentration, cone geometry and operating pressure. Absorption efficiency range from 95–100% with effluent concentrations from 30 to 90 mg/L. Commercial units are available that transfer from 0.2 to 4.9 kg/hr of oxygen per hour (0.4 to 10.8 lbs) at 25 mg/L, at flow rates from 170 to 2,300 Lpm (45 to 600 gpm) (see Figs. 10.8 and 10.9).

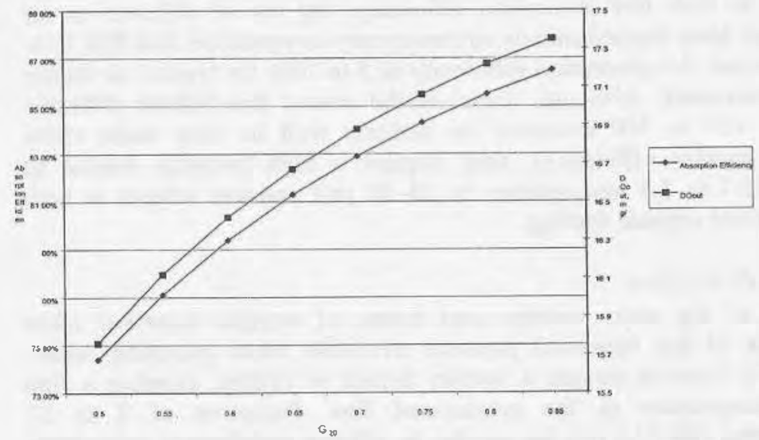


Figure 10.7 Absorption efficiency and effluent DO as affected by gas transfer coefficient, G_{20} (Arbitrary set of model inputs were: Y_2 = 9.5 mm; Y_3 = 13 cm; Y_4 = 61 cm; Y_1 = 7.5 cm; Temp = 20.0°C; Top area = 0.1 m^2; Active Hole Area = 10.0%; Chambers = 10; G/L = 0.01; DO_{in} = 6.0 mg/L; DN_{in} = 14.0 mg/L; DCO_2= 0.0; Pressure = 760.0 mm Hg; Oxygen fraction in inlet gas = 0.99).

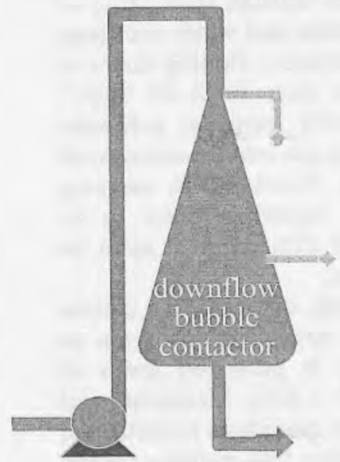


Figure 10.8 Schematic of a Downflow Bubble Contactor.

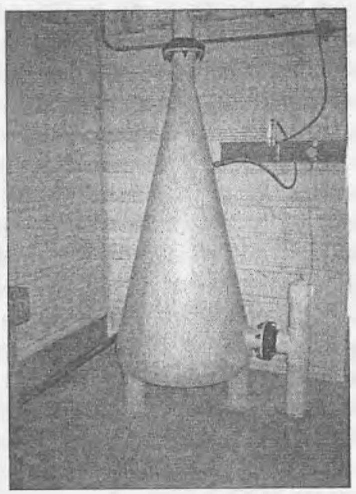


Figure 10.9 Downflow Bubble Contactor.

DIFFUSED AERATION (AIR STONES)

Due to their low absorption efficiency, the use of diffusers or air stones has been limited mainly to emergency oxygenation and fish live-haul systems. An absorption efficiency of 5 to 10% for typical air stones can be assumed. Although some of the recent fine-bubble diffusers (bubbles 100 to 500 microns) do perform well in deep tanks (50% oxygen transfer efficiency), they require a high pressure source of oxygen (1.7 to 3.4 atmospheres or 25–50 psi) and are subject to both chemical and organic fouling.

OXYGEN INJECTION

One of the most widely used forms of oxygen injection takes advantage of the increased pressure available when pumping water. Oxygen is injected though a venturi nozzle or orifice, creating a fine bubble suspension in the pressurized line. Pressures of 2 to 22 atmospheres (30–235 psi) are needed to achieve satisfactory absorption, with contact times of 6–12 seconds. Absorption efficiency ranges from 15 to 70% with effluent DO concentration form 30–50 mg/L.

10.4 DEGASSING: CARBON DIOXIDE (NITROGEN)[c]

Carbon dioxide is introduced into the water through respiration of the fish and bacteria. As stocking densities increase and water exchange rates decrease, dissolved carbon dioxide will become a limiting factor to production. When stocking densities were less than 30 to 60 kg/m^3, conventional aeration systems would generally provide sufficient removal of CO_2 in the process of transferring oxygen into the water with airstones and surface agitation or water falls. However, as carrying capacities have increased to 100 kg/m^3 and higher in order to be economically competitive, pro-active control of CO_2 removal must be included in any successful fish production system.

For every mole of oxygen that is consumed, one mole of carbon dioxide is produced. Alternatively, on a mass basis, for one gram of oxygen consumed; 1.38 g of carbon dioxide is produced (ratio of molecular weights is 44 for CO_2 to 32 for O_2 or 1.375). Measuring and calculating the concentration of dissolved carbon dioxide is complicated in that CO_2 is part of a chemical equilibrium system that includes carbon dioxide (CO_2), carbonic acid (H_2CO_3), bicarbonate (HCO_3^-) and carbonate (CO_3^-) ions. As a result, the concentration of any of these compounds is pH dependent, and to accurately determine their concentrations, values for pH, alkalinity, salinity, and temperature must be known. Figure 10.10 shows the effect of pH on the concentration of CO_2 as a function of pH for alkalinities of 50, 100 and 150 mg/L. Note that CO_2 concentration is proportional to alkalinity.

[c] Software to predict CO_2 removal is provided at www.bee.cornell.edu/aqua.

Carbon dioxide is toxic to fish because it reduces the capacity of the blood to transport oxygen. As the CO_2 concentration in the water increases, so does the blood CO_2 level. The presence of CO_2 in the blood reduces the affinity of hemoglobin to oxygen molecules, a condition often referred to as the Bohr effect or reduces the maximum capacity to bind oxygen (the Root effect) (Wedemeyer, 1996). The safe operating levels of CO_2 depend on the species, development stage, and overall water quality. For tilapia and striped bass, concentrations of CO_2 as high as 60 mg/L have shown no adverse affect. For trout, values of 9–30 mg/L are generally assumed safe. Carbon dioxide may also affect the fish and the biological filter's bacteria by reducing the overall system's pH.

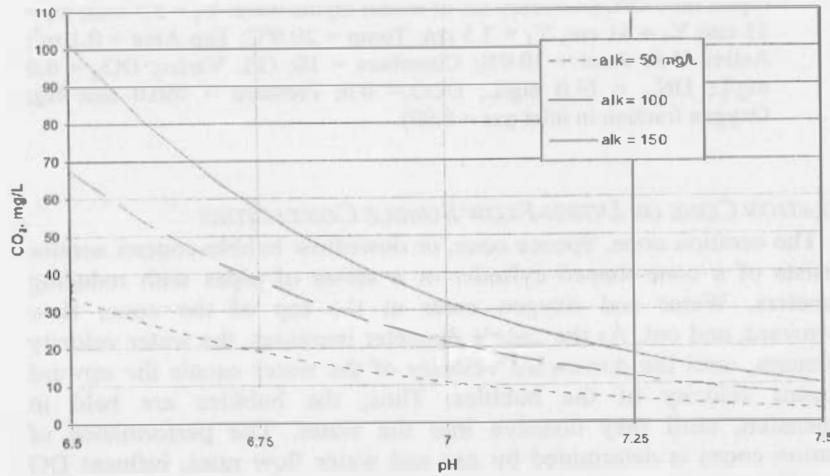


Figure 10.10 CO_2 Related to pH for alkalinities of 50, 100, and 150 mg/L.

Removal of dissolved carbon dioxide is easily done through a gas exchange process, but accurately predicting the removal rate is very difficult. Unlike dissolved oxygen, carbon dioxide is part of a complex equilibrium system, where as CO_2 is removed, a shift in the carbonate carbon equilibrium occurs that also affects pH and CO_2 concentration (see Chapter 2 for much more detail). For example, bicarbonate (HCO_3^-)

acts as a reservoir of carbon dioxide, replenishing dissolved CO_2 as it is removed from solution. In addition, since the concentration of CO_2 in intensive systems is typically more than 20 to 100 times the ambient saturation concentration, the off gassing of CO_2 has a significant effect on the contacting gas. What this means in practice is that air bubbling through or passing over a gas exchange system is quickly saturated with CO_2.

Thus, large volumes of air per unit volume of water are required to strip off CO_2. For example, in normal aeration, gas to liquid ratios (G/L) are usually less than 3 to 1 (3 unit volumes of gas to every unit volume of water). In oxygenation, the G/L ratio is between 0.003:1 to 0.05:1. But for CO_2 stripping, the G/L ratio is between 5:1 to 20:1. Using a packed column and gravity flow of water to supply the packed column is one of the easiest ways to strip water of CO_2.

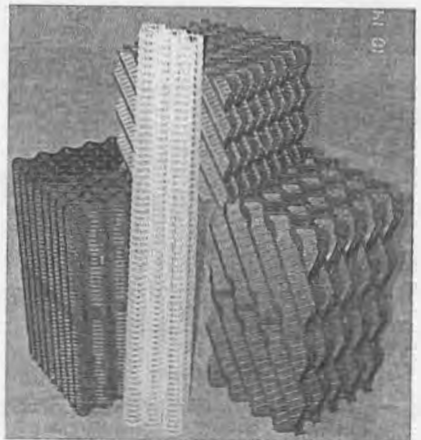


Figure 10.11 Two types of packing media for air-stripping columns.

Carbon dioxide removal as a separate water treatment process may be an issue when groundwater resources are used and has become especially important with the relatively recent intensification of aquaculture systems. While air-contact aeration processes remove dissolved carbon dioxide, their primary purpose is often considered the addition of oxygen. Air-contact aeration works well in low intensity aquaculture systems that do not use oxygenation and generally have sufficient water exchange and/or aeration to keep carbon dioxide from

accumulating above safe levels. However, carbon dioxide problems have appeared in intensive systems using pure oxygen absorption because of the relatively low specific water exchange rates used (rate of make-up water flow per mass of fish in the system) and the fact that pure oxygen systems typically do not remove substantial quantities of carbon dioxide. Under intensive aquaculture conditions, the accumulated concentration of dissolved carbon dioxide in the culture tank will not be limiting (with no aeration or pH control) when the cumulative dissolved oxygen consumption is less than 10 to 22 mg/L, depending upon pH, alkalinity, temperature, salinity, and the species and life stage (Colt et al. 1991). Once this cumulative oxygen consumption level is reached, the water flow cannot be used again unless it is air-stripped of carbon dioxide, or a chemical is added to reduce carbon dioxide concentrations. Therefore, carbon dioxide control in water reuse systems may require aeration for stripping, the use of chemicals to adjust pH, or a combination of both methods.

The open packed column for carbon dioxide removal is a vertical tower filled with plastic packing similar to a pure oxygen packed column absorber. Packing media appropriate for this application is shown in Fig. 10.11. In operation influent water is introduced at the top of the column and is distributed uniformly over the plastic packing with a perforated plate, spray nozzle, or spray bar. Water flows downward through the packing and is broken up creating a large gas-liquid interfacial area for gas transfer. To overcome the potential for clogging or biofouling of plastic packing alternative water breakup approaches have also been utilized. It is common for the stripping column to have no packing or just a few splash screens to break the water up as it travels through the column (see Fig. 10.12). If plastic packing is used provisions are made to allow for particulates to pass through the media by using high porosity packing or packing that has an unobstructed downward path through the column.

Taking advantage of the water breakup created by the packing media and passing large volumes of air through the column achieve carbon dioxide removal. Fresh air has a low gas phase concentration of carbon dioxide (it used to be 350 ppm and it is now over 380 ppm...) and is suitable for providing an adequate driving force for carbon dioxide gas transfer. Carbon dioxide is degassed from the water being treated and is carried out of the column with the fresh air blown through. In contrast with pure oxygen devices that use volumetric gas flow to water flow (G/L ratio) ratios of 0.3–5%, carbon dioxide stripping columns use G/L ratios of 500-2,000%. Low horsepower, high volume, ventilation-type

air blowers provide the large amounts of airflow required for efficient carbon dioxide removal.

Stripping column design heights are generally limited to 1.0 to 1.5 m (3.3–4.9 ft) because of the diminishing returns on removal performance that correspond to packing heights greater than 1.5–2.0 m (4.9–6.6 ft). Hydraulic loading for carbon dioxide stripping columns are reported to range between 0.61–2.51 Lpm/m^2 (25–102 gpm/ft^2). However, values towards the lower end are recommended to reduce the back-pressure on the blowers when the column design calls for packing depths exceeding 1 m (3.3 ft) (Summerfelt, et al. 2000).

Figure 10.12 shows two types of counter-current air-stripping columns with media or if solids are a problem, splash screens.

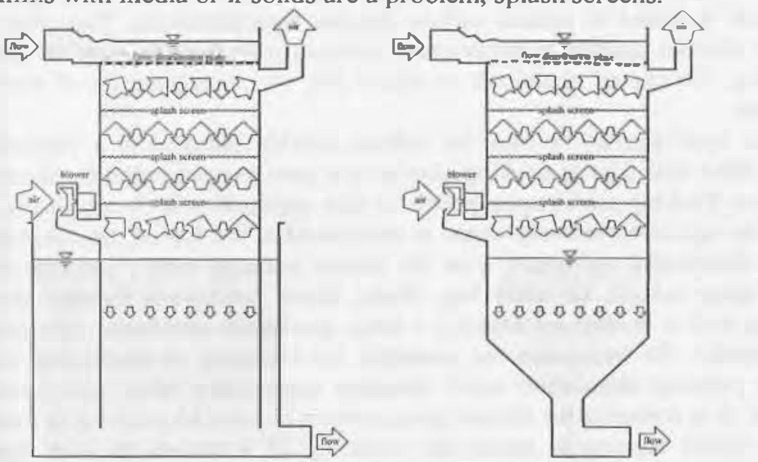


Figure 10.12 Carbon dioxide stripping columns. Air is blown through stripping columns, often counter-current (shown here) to the water falling through splash screens or media. Air-stripping columns can also be constructed within cone-bottom cylindrical tanks (right diagram) to avoid a sump were settleable solids can accumulate (left diagram) (from Summerfelt et al. 2000).

When possible, try to blow the air upwards through the falling water streams to maximize stripping effectiveness. Design of counter current air-stripping columns requires selecting the packing material, determining the packing depth, hydraulic loading rate, volumetric air to water (G/L) ratio and estimated inlet CO_2 concentration. An interactive computer program to assist in the design of these systems is included in the appendix. In general, a CO_2 degasser is designed with a hydraulic fall of between 1 to 1.7 m (3 to 5 ft), a hydraulic loading of 17–24 L/m^{2}s (25–35 gpm/ft^2) a volumetric air-water ratio of from 5:1 to 10:1 and high

porosity packing. Summerfelt et al. (2000) demonstrated the impact of increasing G/L ratios and column packing height on the percent of CO_2 removed per single water treatment pass (see Fig. 10.13).

Many times the primary interest of the design engineer is the impact of G/L ratio on the relative effectiveness of a particular device. For example, if a lower G/L ratio were used, what fraction of the maximum attainable removal would be attained for that particular device. The results shown in Fig. 10.13 were re-plotted in this fashion to show what percentage of the removal was attained as impacted by increasing G/L ratio (see Fig. 10.14).

Figure 10.13 Packing height and G/L ratio effect on percent of influent CO_2 removed.

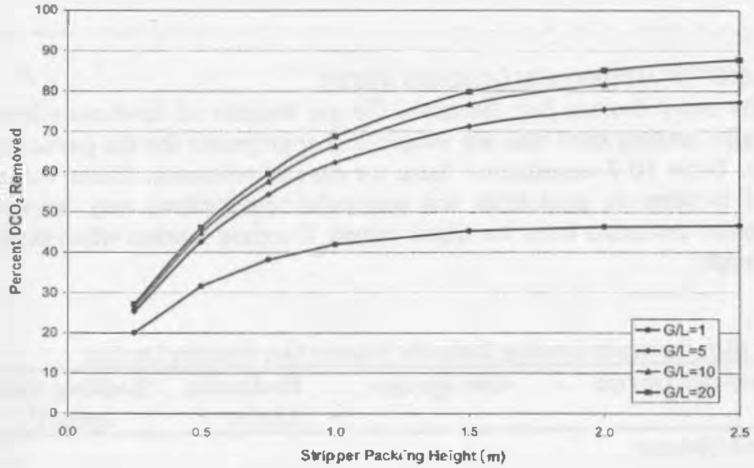


Figure 10.14 Effect of G/L Ratio on the relative CO_2 removed (G/L = 20 being defined as 100% of maximum achievable) for packed columns of different heights.

Where nutrient levels in the water may be high, stripping towers without packing media may be preferred to reduce maintenance time and variability in performance due to biofouling. The CO_2 removal percent as affected by fall height from a flooded plate to a receiving pool (similar to an LHO, except there is only a single chamber) is shown in Fig. 10.15. Note that the stripper column with the larger drop height requires a greater G/L ratio to remove the same amount of CO_2 as the shorter unit. A generic graph (see Fig. 10.16) is presented similar to Fig. 10.14 to show the relative impact of G/L ratios on removal fraction of the dissolved CO_2 for the two tower stripping heights. The LHO program given in the appendix can be used to reconstruct these curves by setting the oxygen feed purity at 20.9%, i.e., air.

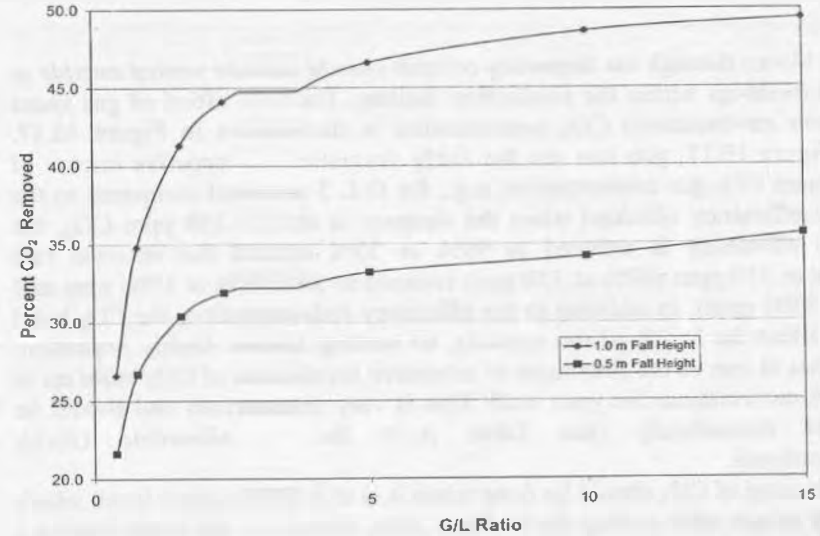


Figure 10.15 Effect of G/L ratio on the % of CO_2 removed as affected by height of fall in a stripping chamber without packing.

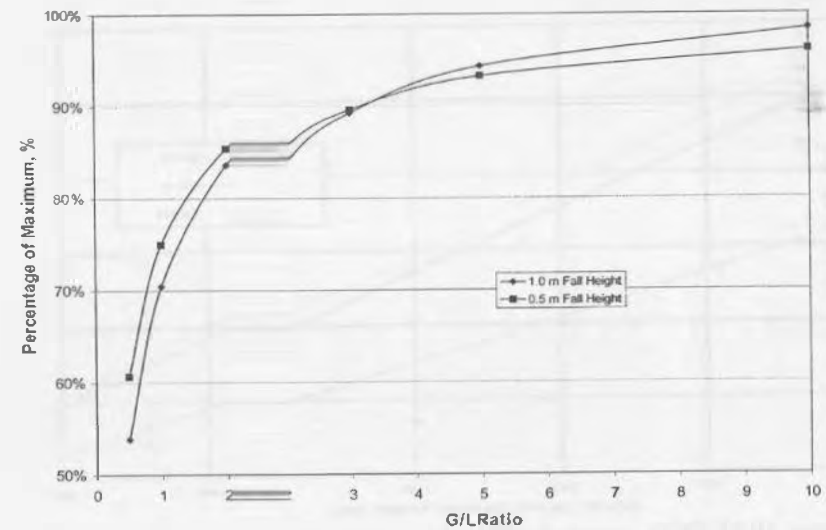


Figure 10.16 Effect of G/L Ratio on the relative CO_2 removed (G/L = 15 being defined as 100% of maximum achievable) for stripping columns with no packing (fall height of 0.5 or 1.0 m).

Air blown through the degassing column ***should be vented outside to prevent build-up*** within the production facility. The effect of gas space (the room environment) CO_2 concentration is shown in Figure 10.17. From Figure 10.17, you can see the fairly dramatic ***negative*** impact of background CO_2 gas concentration, e.g., for G:L 5 and compared to the removal efficiency obtained when the airspace is at 350 ppm CO_2, the removal efficiency is reduced to 90% or 33% of the removal rate obtained at 350 ppm (60% at 350 ppm reduced to 54% at 1000 ppm and 18% at 5000 ppm). In addition to the efficiency reduction, the CO_2 build up can affect the health of the workers, so venting is doubly important. Headaches is one of the first signs of excessive levels of CO_2 build up in the work environment for your staff. This is very serious and should be corrected immediately (see Table A-15 for allowable OSHA concentrations).

Degassing of CO_2 should be done when it is at it highest level, which normally occurs after exiting the biofilter. Also, since the water leaving a degassing column is elevated to within 90% of saturation, additional oxygenation should occur after degassing and preferably just before the water enters the production tanks.

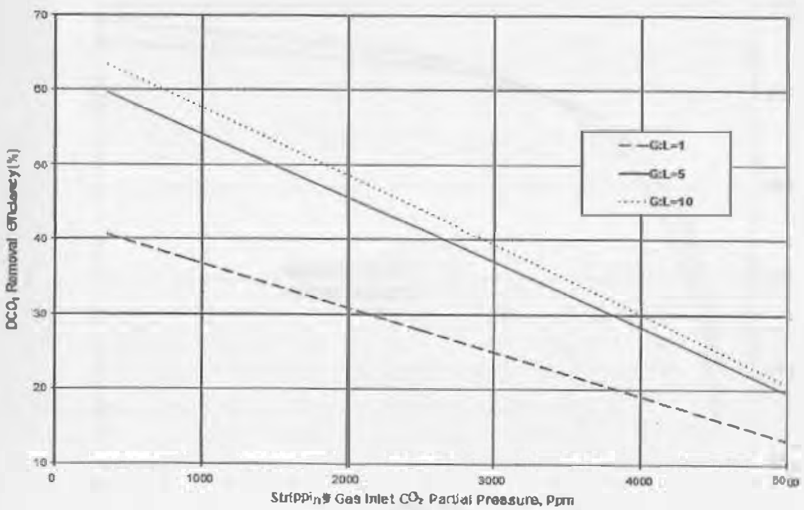


Figure 10.17 DCO_2 removal efficiency versus stripping gas inlet CO_2 partial pressure for a 1 m tall packed column filled with 2 inch Norpac. Given: Inlet DCO_2=13.6 mg/L; Water temp=14.4°C; BP=750mm Hg; Alkalinity=196 mg/L; Hydraulic loading=20 kg/m^{2}s

SUMMARY OF HYDRAULIC LOADING RATES

The many devices just discussed for gas transfer all have associated hydraulic loading rates that are considered appropriate for the particular device. Table 10.7 summarizes these for ease of reference. These values should be seen as guidelines and particular applications may warrant substantial deviation from the listed values. Exercise caution when doing so, though.

Table 10.7 Hydraulic Loading Rates for Various Gas Transfer Devices

Gas Transfer Device	Gas Species	Hydraulic Loading Rate L/m^{2}s	gpm/ft^2
Packed Columns			
Sealed	oxygen	<166	<244
High pressure	oxygen	45–246	66–361
Pure oxygen, low pressure	oxygen	73	107
Open to atmosphere	carbon dioxide	17–68	25–100
Spray Tower	oxygen	35–95	51–140
Low Head Oxygenator (LHO)	oxygen	34–68	50–100
Down Flow Bubble Contactors (Speece cone)			
at inlet	oxygen	1,800	2650
at outlet (to keep bubbles in)		150	220
U-Tubes	oxygen	2,000–3,000	2,940–4,410

INSTALLATION AND SAFETY CONCERNS

The handling and use of pure oxygen can be ***hazardous***. Almost all substances will burn and in some cases explosively in an oxygen enriched environment. There have been rumors of used air stones exploding, when transferred to pure oxygen systems, when the organic

residue oxidized. Special care and materials should be used in the construction and installation of pure oxygen systems. Stainless steel or copper tubing is recommended for oxygen lines, and both piping and fittings should be cleaned of grease and oil, which can present a fire hazard. Oxygen piping is special order material that comes sealed to prevent organic contamination that could later lead to a gas explosion. You can not be too careful in dealing with your oxygen piping installation. Petroleum products should never be used on hardware that handles or contacts pure oxygen. Fittings and valves that have been specially cleaned and packaged for pure oxygen applications are available. Liquid oxygen is very, very cold and installation and servicing of storage containers should be left to the professionals.

10.5 Design Example – Aeration/Oxygenation

Because the stocking densities for the Omega fish production system is at a low-density (less than 40 kg/m^3 or 0.33 lb/gal), for most of the growout period only aeration (using air, no pure oxygen) in the fry/quarantine tanks, and the fingerling systems is required. It would only be during the final few weeks of growout that the system biomass and feed rate could be high enough to demand supplemental pure oxygen. There would be two options to provide that oxygen, using micro diffusers in the individual tanks or a Speece cone in one of the return lines to the system. Fine-bubble micro diffusers create what can only be described as an oxygen bubble cloud in the water column and have good oxygen transfer efficiencies, especially in deep tanks. They do require a high pressure source of oxygen (1.7 to 3.4 atmospheres or 25–50 psi), which makes them a good match for either a pressure swing adsorption (PSA) or a vacuum swing adsorption (VSA) unit or for an on-site Dewar's type storage containers. For large commercial operations (exceeding ~200 ton/year of production), leasing oxygen tank systems from a national supplier will probably be one of the most viable options. The flow rate of oxygen to the growout tank can then either be directly controlled by an operator to maintain some set point by monitoring the DO in the tank on a regular basis, or using a continuous monitoring system with a setpoint control point and a relay operated solenoid valve. Remember, under intensive conditions, you can lose all your fish in 15 minutes, so continuous monitoring is something you must employ (see Chapter 13 Monitoring and Control for additional discussion).

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LIST OF SYMBOLS

A	Total hole area over the entire LHO, m^2
A_O	Constant used for regression in water vapor equation, dimensionless
C_d	Discharge coefficient to predict velocity through an orifice, dimensionless
C_i	Concentration of gas species "i", mg/L
C_{in}	Influent dissolved concentration of a gas species, mg/L
$C_{meas,i}$	Measured concentration of gas "i" in water, mg/L
C_{out}	Effluent dissolved concentration of a gas species, mg/L
$C_{s,i}$	Dissolved gas saturation concentration for species "i", mg/L
DN	Dissolved nitrogen concentration, mg/L
DO	Dissolved oxygen concentration, mg/L
E	Gas transfer efficiency, %
F	Gas mean residence time for one flushing to occur for a chamber, hr
g	Acceleration due to gravity, 9.81 m/s^2
G_{20}	Overall mass transfer coefficient at 20°C, dimensionless
G_T	Overall mass transfer coefficient at a specific temperature T°C, dimensionless
G/L	Gas to liquid ratio, both being on a volumetric basis, dimensionless
h	height above sea level, m
HL	Hydraulic loading, L/m^2 per s
J_i	Constants used to calculate gas partial pressure due to a specific gas "i" dissolved to some concentration in water, dimensionless
P_{BP}	Barometric pressure, mm Hg
P_i^l	Partial pressure of gas "i" in liquid form, mm Hg
P_i^g	Partial pressure of gas "i" in gas phase form, mm Hg
P_{TG}	Total gas pressure, mm Hg
P_{WV}	Water vapor pressure, mm Hg
Q	Rate of gas flow, kg/time
R	Gas flow rate into an LHO chamber, L/hr
$R_{pressure}$	Resistance to gas transfer
$S_{\%}$	Percent saturation of a particular gas, %
T	Temperature, °C
TGP	Total gas pressure, mm Hg
V	Volume of single mixing chamber in an LHO, L
Y_1	Hydraulic head over flooded plate, cm
Y_2	Hole diameter, mm
Y_3	Pool depth, cm
Y_4	Fall height of water from plate to receiving pool, cm
Z	Packing height of media, m
Z_{tower}	Height of an enclosed spray tower, m
α	Ratio of field water G_{20} / clean water G_{20}, dimensionless
β_i	Bunsen Coefficient for gas species "i", L/L-atm
K_i	Ratio of the molecular weight to volume (mg/mL)
X_i	Mole fraction of gas (dimensionless)

CHAPTER 11

OZONATION AND UV-IRRADIATION[1]

11.0 INTRODUCTION

Bacterial and viral diseases create serious problems in semi-intensive and intensive aquaculture. Use of surface water in flow-through systems represents a risk of contamination by introducing waterborne fish pathogenic microorganisms. Such contamination results in heavy losses in aquaculture worldwide, and has also limited the progress in commercial farming of new aquacultural species. Some commercial operations may be required to disinfect their discharge waters before release into the aquatic environment. Of serious concern are dependable means of controlling pathogens present in the inlet water. Disinfection by ozonation or UV-irradiation are two methods often applied in aquaculture. It is important to distinguish between disinfection of makeup waters (low organic loads) and recirculating aquaculture system (RAS) waters. Both applications are discussed in this chapter. Disinfection by ozonation and UV-irradiation are also used in other aquacultural applications, e.g., reducing or eliminating potential pathogens associated with live prey such as rotifers in marine larval production systems, and surface disinfection of fish eggs. Other methods of disinfection are also discussed at the end of this chapter.

11.1 UV IRRADIATION

Natural and artificial UV light (wavelength of 190–400 nm) may damage microorganisms by directly and indirectly altering nucleic acids. Direct damage is due to absorption of irradiation by DNA with resulting formation of photoproducts. DNA absorbance is high in the UV-C range (190–280 nm), but falls more than three orders of magnitude in the UV-B range (280–320 nm), is and negligible in the UV-A band (320-400 nm) (Miller et al. 1999). The DNA-damaging effect of UV-C is utilized

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in bactericidal lamps. The low-pressure mercury-vapor lamp emits approximately 85% of its energy output as monochromatic light at a wavelength of 253.7 nm, which is within the optimum wavelength range of 250 to 270 nm for bactericidal effects. Solar UV-B irradiation is responsible for both direct and indirect DNA damage, while UV-A produces only indirect damage. Only direct DNA damage is repairable by photoreactivation.

The UV-C damage often results from dimerization of two pyrimidine molecules. Cyclobutane pyrimidine dimers and pyrimidine-pyrimidone (6-4) photoproducts are the two major classes of photolesions formed by direct DNA absorption of UV-C irradiation (Friedberg et al. 1995). Once the pyrimidine residues are covalently bound together, replication of the nucleic acid is blocked or results in mutant daughter cells unable to multiply (Stover et al. 1986). The moderate energy level of UV irradiation leaves no toxic residuals in the treated water. Although chemical compounds can be altered by the radiation (Gjessing and Kallqvist, 1991; Lund and Hongve, 1994), the UV doses used for disinfection are too low to generate significant amounts of photoproducts (Oliver and Carey, 1976; de Veer et al. 1994). This non-toxicity is of crucial importance when UV is the method of choice for influent disinfection in aquacultural facilities.

UV irradiation units are commonly installed in smolt farms for sea and freshwater disinfection, and are also used for bacteriological control in recirculation systems (Rosenthal, 1981). However, before the UV dose can even reach the target organism, it must be able to transmit through the water. UV applications in highly turbid water as is often the case in RAS will be totally ineffective since the transmission into the water column is very minor, thus killing almost no organisms. Therefore, the lowest expected UV transmittance of the process water should be established and used to predict how much UV intensity must be generated to transmit the desired UV dose through the water between the target organism and the light source.

Ultraviolet filters can be built as nonpressurized open channel units (see Fig. 11.1) or as pressurized tube-and-shell units. The UV bulbs are usually contained within quartz sleeves to allow submergence in the process flow. The quartz sleeves must be kept clean to maintain transmittance. Using UV light does not produce toxic residuals or form byproducts that pose a risk to aquatic organisms.

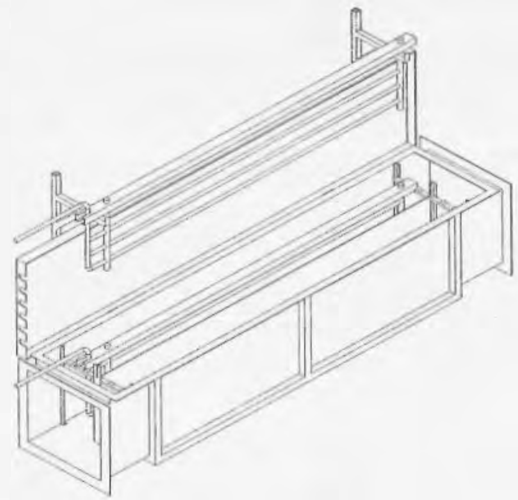

Figure 11.1 A horizontal UV channel filter in position mode for bulb removal. Drawing courtesy of PRAqua Technologies, Ltd., Nanaimo, British Columbia (from Summerfelt et al. 2001).

UV Light Output

UV light output strength continually deteriorates from the time you first turn on the machine. Assume in general a 3% per month decrease in output strength or 40% per year. UV output strength is also affected by air temperature. The UV output ratings are 100% at 100°F (38°C); at 32°F (0°C), the output strength is only 10% of the 100°F (38°C) rating. This problem can be addressed by using quartz sleeves that creates a warm air pocket around the UV bulb and this maintains output strength near its maximum potential. The quartz sleeve will reduce output by 5% just due to transmissivity of the sleeve. Bulb replacement in UV units can be of considerable expense, particularly when compared to other forms of sterilization and disinfection. As a rule of thumb, change UV bulbs at least once annually.

"Rule of Thumb"
Change UV bulbs at least once a year.

Application Example of UV

Problem

Define a UV unit to provide a minimum lethal dose (MLD) for an organism that requires 100 mWs/cm^2 in an incubator system with a water volume of 1,000 L.

Solution

Select a UV unit from a commercial manufacturer. Such units are rated on maximum flow rate through the unit and their dosage output. Assume you select a unit that is rated at 10 L/min with an output of 40 mWs/cm^2. Now, calculate the allowable flow rate to achieve the specified MLD for the organism we are attacking:

$$Q\frac{L}{\min}\cdot 100 mWs/cm^2 = 10\frac{L}{\min}\cdot 40 mWs/cm^2$$

$$Q = 4.0\ \frac{L}{\min}$$

Thus, although the unit is rated for a flow of 10 L/min, you can only operate the unit at a flow rate of 4 L/min. Thus the 1,000 L incubator volume will only be turned over once every 250 minutes or about 6 times per day. This is acceptable. There is clearly judgment involved in determining if you are cycling your water through a UV unit often enough. The turnover has to be sufficiently fast so that organisms do not generate faster than they are being killed off.

11.2 OZONATION

Ozone has seen wide use in aquaculture because it has a rapid reaction rate, produces few harmful reaction by-products in freshwater and oxygen are produced as a reaction end-product. Ozone is an extremely reactive oxidant and a very effective bactericide and viricide. Ozone can also be used to achieve water quality improvements by microflocculating fine particulate matter (making particles that are easier to settle or filter) and oxidizing nonbiodegradable organic molecules (creating smaller and more biodegradable molecules), nitrite, and refractory organic molecules (reducing water color) (Summerfelt and Hochheimer, 1997; Summerfelt et al. 1997).

Application of ozone to aquaculture requires ozone generation, ozone transfer into solution, contact time for ozone to react and disinfect, and possibly ozone destruction to ensure that no ozone residual makes it into the culture tanks (Summerfelt and Hochheimer, 1997).

Ozone Generation

Ozone must be generated on-site. The most efficient method is by the electric corona discharge technique, which involves the passage of oxygen gas, or air, across a gap of narrowly spaced electrodes under high voltage, Fig. 11.2. According to Masschelein (1998), effective ozone generation depends upon the composition of the feed gas, e.g., feed gas impurities, particulates, moisture, and pressure of the feed gas, efficiency of dielectric cooling, characteristics of the electrical current, concentration of the ozone produced, and dielectric design. Protecting the dielectrics within the corona discharge cells requires a feed gas supply that is dry, free from particulate matter and coalescible oil mists, and contains less than 15 ppm hydrocarbons (Dimitriou 1990; Masschelein, 1998). Also, some ozone generators may require some nitrogen gas impurity (>0.5% nitrogen) within the oxygen feed gas in order to achieve maximum ozone production efficiency. To meet this level of nitrogen contamination, some liquid oxygen supplies may require adding a small quantity of nitrogen to the feed gas before it enters the ozone generator.

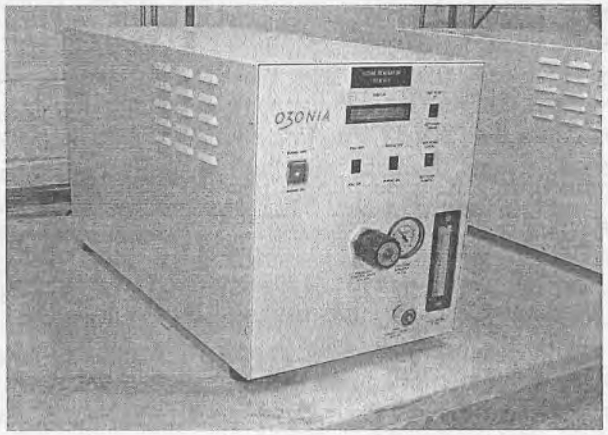

Fig. 11.2 Electron corona discharge ozone generator.

Purified oxygen is already used to maximize carrying capacity within many intensive aquaculture systems. Corona discharge generators using

purified oxygen feed gas require about 10 kWh of electricity to produce 1.0 kg of ozone (Masschelein, 1998). However, ozone production with an air feed gas is 2–3 times less energy efficient than using purified oxygen feed gas (Masschelein, 1998). Also, generating ozone in oxygen feed gas can produce a 10–15% (by weight) concentration of ozone, which nearly doubles the concentration of ozone that can be generated using air as the feed gas. The relatively high concentrations of ozone can be generated to reduce the overall mass of oxygen required to supply ozone. Yet, it is less energetically efficient to produce ozone concentrations of 10–15% (by weight) than to produce ozone concentrations of 4–6% (Carlins and Clark, 1982). Taking all of this into account, ozone production can be optimized according to the demands of the aquaculture system and economic considerations of feed gas cost and energy usage.

OZONE TRANSFER

Ozone is generated within either air or oxygen feed gas and this ozone/oxygen gas flow must be transferred into water for microbiological inactivation or other oxidative purpose. The ozone gas flow can be co-transferred into the water using any of the typical oxygen transfer devices (Summerfelt and Hochheimer, 1997), which have been discussed in Chapter 10 of this book. Effective transfer of ozone gas into water is important because the cost of producing ozone is not insignificant, especially if the ozone is carried within purified oxygen feed gas that was either purchased or produced on site.

The rate of ozone transfer and the subsequent rate of ozone decomposition depend upon the efficiency of the contacting system used and the rates that ozone reacts with constituents within the water. The rate that ozone reacts depends on the type and concentration of constituents within the water. Rapid reaction with oxidizable inorganics and organics will maintain a low apparent equilibrium concentration of ozone within the liquid film and increase the rate of ozone transfer. The driving force for ozone transfer is maximized when the ozone absorbed is rapidly consumed by reaction with constituents within water. In fact, when ozone reacts very fast, ozone decomposes at the gas surface and no molecular ozone is transferred into the water (Bablon et al. 1991).

Ozone transfer units that have a continuous liquid-phase, i.e., units that disperse gas bubbles within a liquid, -- such as U-tubes, Speece cones, Fig. 11.3, aspirators, bubble diffusers, and enclosed mechanical surface or subsurface mixers -- provide both ozone transfer and some reaction time. Ozone transfer units that have a continuous gas-phase, i.e., units that disperse liquid drops and films within a gas, such as spray columns, packed columns, and multi-stage low head oxygenators, Fig. 11.4, provide efficient transfer but very little time for reaction (Summerfelt and Hochheimer, 1997). Continuous gas-phase transfer units are best suited for use in situations that normally require the transfer of the maximum amount of ozone in the shortest time for economical fixed and variable costs. On the other hand, continuous liquid-phase transfer units are usually selected for situations where reaction is rate limiting and an ozone residual must be maintained for a specific length of time (Bellamy et al. 1991).

Most ozone contactors rely on continuous liquid-phase units that bubble ozone into the liquid (Bellamy et al. 1991). High column bubble diffusers are frequently used for aquacultural applications and can achieve more than 85% ozone transfer to the liquid phase (Helge Liltved, personal communication). These units are particularly well suited to situations where reaction is rate limiting and an ozone residual must be maintained for a specific length of time, such as during disinfection. Speece cones, Fig. 11.2, and U-tubes are also being used to efficiently and rapidly transfer ozone/oxygen feed gas within RAS, where oxidation of nitrite and organic matter are the primary goals of ozonation (not disinfection).

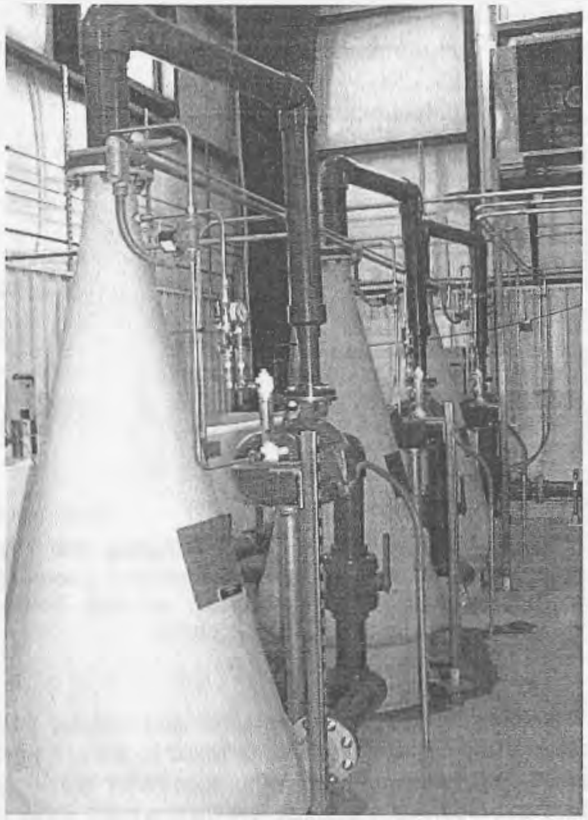

Figure 11.3 An ozone/oxygen feed gas is injected into water within three Speece cones (plumbed in parallel for redundancy and variable flow requirements). System shown is used to disinfect 400–2400 L/min of surface water at the US Fish and Wildlife Service's Lamar National Fish Hatchery (Lamar, PA). Ozone transfer efficiency is typically >99% efficient within these cones when operated at 8–10 psig (0.5 to 0.7 atm) (Summerfelt, personal communication).

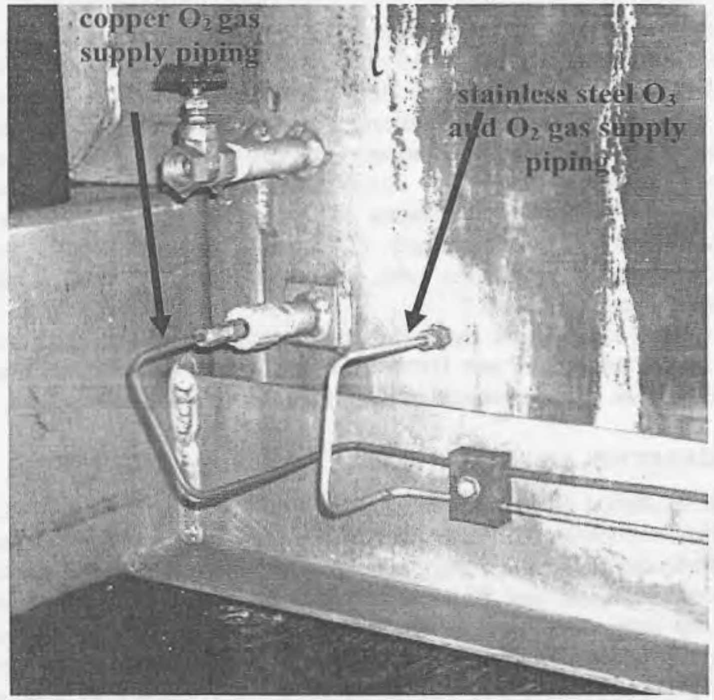

Figure 11.4 An ozone/oxygen feed gas is injected into water within an LHO used to oxygenate/ozonate 4800 L/min of recirculating water in a system at the Freshwater Institute (Shepherdstown, WV).

Ozone transfer within continuous gas-phase units is not as common as within continuous liquid phase units (Bellamy et al. 1991). When ozone transfer has been reported within continuous gas-phase units, they are mostly packed columns. However, continuous gas-phase units can be designed to efficiently transfer ozone within relatively smaller vessels. Ozone transfer efficiency was 100% in the LHO (Low Head Oxygenator) units evaluated in the recirculating system at The Freshwater Institute. In this system, complete ozone transfer occurred because ozone is 13 times more soluble than oxygen in water according to Henry's law; short circuiting in the gas phase within the LHO was prevented by breaking the chamber into eight separate compartments; gas residence times within the LHO chambers were about 45 min; and, there was nitrite and dissolved and suspended organic material in the water that rapidly reacted with the dissolved ozone (Summerfelt and Hochheimer, 1997).

Ozone transfer into RAS is sometimes accomplished using the same gas transfer unit that is used for oxygen supplementation. This can be done if the transfer unit is fabricated from ozone resistant material (Bullock et al. 1997). In these situations, adding ozone to a recirculating system that is already using purified oxygen only requires installation of an ozone generator and the accompanying ozone distribution, monitoring, and control mechanisms (Summerfelt and Hochheimer, 1997). All of the other necessary equipment (oxygen supply and distribution system, gas transfer units, and control mechanisms) would already be in place, Fig. 11.4.

The off-gas discharged from the transfer unit will contain some ozone if ozone transfer is not 100% efficient. These ozone containing off-gas discharges must be treated to destroy remaining ozone.

Ozone Reaction and Inactivation of Fish Pathogens

Ozone oxidation can kill microorganisms, but requires maintaining a certain dissolved ozone concentration in the water for a given contact time. Disinfecting efficiency depends upon the product of the ozone residual concentration and its contact time. An ozone contact vessel provides the time necessary for the ozone residual to react with and inactivate pathogenic microorganisms. Disinfecting waters may require maintaining a residual ozone concentration of 0.1–2.0 mg/L in a plug-flow type contact vessel for periods of 1–30 min, depending upon the target microorganism (Wedemeyer, 1996). In commercial aquaculture applications, it is extremely difficult to maintain residual concentrations above 1 mg/L; above 2 mg/L is almost impossible with conventionally available equipment. An example system that provides ozone contact time in a two reactor sequence vessel with residual removal is illustrated in Fig. 11.5. The primary ozone treatment components are located on the process pipeline immediately after ozone gas has been transferred into the water. First, the right hand vessel provides 10 minutes (at 1600 L/min flow) of plug flow contacting to achieve disinfection. Next, the middle vessel provides 20 minutes (at 1600 L/min flow) of plug flow contacting to achieve further disinfection and ozone residual destruction. Finally, the left hand vessel (a counter-current, forced-ventilation column) strips any minor ozone residuals and elevated dissolved oxygen levels immediately before the flow is piped to the fish culture systems.

Ozone exposure experiments with bacterial cells have indicated changes in membrane structure which lead to leakage of protein and nucleic acid, and also lipid oxidation, while the intracellular components, protein, and DNA, remain intact (Komanapalli and Lau, 1996). By prolonged ozone exposure, cell viability is reduced with a more

significant increase in lipid oxidation and protein and nucleic acid leakage.

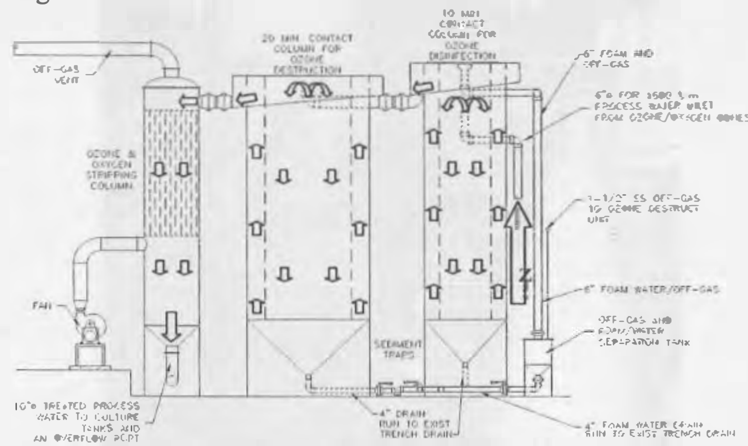

Figure 11.5 Ozone treatment system for disinfecting 400–2400 L/min of surface water at the US Fish and Wildlife Service's Lamar National Fish Hatchery (Lamar, PA). Drawing courtesy of Oak Point Associates, Biddeford, Maine (from Summerfelt et al. 2001).

It has been demonstrated that bacteria and viruses pathogenic to salmonids are highly sensitive to residual ozone in water (see Tables 11.1 and 11.4). Dose-response estimates of this sensitivity are fairly precise in demand-free water (inorganic buffers, distilled water), indicating 99.9% inactivation or more in the 0.01–0.10 mg/L residual concentration range. In batch experiments with natural water exerting an ozone demand, residual concentration tends to drop rapidly, making reliable dose-response estimates more complicated. As a rule, higher residual concentrations, i.e., 0.1–0.2 mg/L in natural sea-, brackish-, and freshwater, and 0.3–0.4 mg/L in fish farm effluents, seem necessary to obtain the legally required inactivation level. However, new results indicate that some viruses, including the infectious pancreatic necrosis virus (IPNV) and Atlantic halibut nodavirus (AHNV), are more resistant to ozonated seawater than previously anticipated (Liltved *et al.* 2006). Considerably higher TRO concentrations and C·T values seem to be required to inactivate these viruses. On the other hand, the infectious salmon anaemia virus (ISAV) was easily inactivated by low ozone dosages.

Table 11.1 Inactivation of Fish Pathogenic Microorganisms in Water by Ozone

Organism	Percent removal	Time (min)	Conc., mg/l	Cxt min × mg/l	Temp. °C	pH	Water	Comments on water quality	References
Bacteria									
A. liquefaciens *A. salmonicida* *P. fluorescens*	99.9	1.2-5.0	0.10	0.12-0.50	20	7	PBS		Colberg and Lingg 1978
A.salmonicida	100	0.5	0.04	0.02	20	7	PBDW	Ozone demand free	Wedemeyer and Nelson 1977
A.salmonicida	99.8	0.33	init. 0.010 res. 0.04		7	7.2	PBS		Liltved and Landfald 1995
A.salmonicida	99.9	1.0	0.3-0.4		7	7.8	Fish farm effluent	TSS 22 mg/l TOC 9.8 mg/l	Liltved and Landfald 1995
A.salmonicida *V. anguillarum* *V. salmonicida* *Y. ruckeri*	99.99	3.0	init. 0.15-0.20 res. 0.05-0.07		9-12	6.3-8.0	Lake, brackish and seawater	Natural waters	Liltved *et al.* 1995
Enterococcus seriolicida, Pasteurella piscicida, V. Anguillarum	99.9	0.9-1.2	0.089-0.177*	0.084-0.186*	25	7.95	Seawater		Sugita *et al.* 1992
Yersinia Ruckeri	100	0.5	0.01	0.005	20	7	PBDW	Ozone demand free	Wedemeyer and Nelson 1977
Yersinia Ruckeri	99.9	3.0	0.10	0.30	20	7	Distilled		Colberg and Lingg 1978

Organism	Percent removal	Time (min)	Conc., mg/l	Cxt min × mg/l	Temp. °C	pH	Water	Comments on water quality	References
Viruses									
IHNV	100	0.5	0.01	0.005	10	7	PBDW	Ozone demand free	Wedemeyer *et al.* 1978
IPNV	100	1.0	0.01	0.010	10	7	PBDW	Ozone demand free	Wedemeyer *et al.* 1978
IPNV	99.99	1.0	0.10-020	0.10-0.20	9-12	6.3-8.0	Lake, sea, brackish	Natural waters	Liltved *et al.* 1995
IPNV	98.4	31	2.5	77.5	5	7.9	Seawater	Natural	Liltved *et al.* 2006
ISAV	99	0.25	0.33	0.08	5	7.9	Seawater	Natural	Liltved *et al.* 2006
AHNV	98	31.5	1.6	50.4	5	7.9	Seawater	Natural	Liltved *et al.* 2006
SJNNV	Loss of infectivity	0.5-2.5	0.1-0.5	0.25			Seawater		Arimoto *et al.* 1996
WSBV	Loss of infectivity	10	0.5*	5			Seawater		Chang *et al.* 1998

*Concentrations recorded as total residual oxidants (TRO)

SJNNV – striped jack nervous necrosis virus

In dose-response trials, two-stage logarithmic inactivation curves have been obtained (Liltved et al. 1995; Liltved and Landfald, 1995). These were characterized by a rapid inactivation initially, followed by a decreasing inactivation rate with exposure time. Such kinetics could be explained by reduced ozone concentration during the course of exposure, and have also been experienced by other investigators, even in ozone demand-free water (Katzenelson et al. 1974; Colberg and Lingg, 1978; Vaughn et al. 1987). In practical ozonation of fish farm influent and effluent water, it is important that the ozone dose is high enough to account for the initial demand, thereby establishing a sufficient residual concentration for the required contact time.

In demand-free water, dissipation of ozone will still be observed due to the demand exerted by the added microorganisms. This demand will depend on type of organism, the preparation, and washing of the inoculum prior to supplementation, and the density of organisms in the ozonated suspension. In natural waters and in waters found within recirculating systems, additional ozone will be lost in reactions with organics and other compounds. According to ozone demand tests on a high quality trout stream water that is being ozone disinfected at the US Fish and Wildlife Service Lamar National Fish Hatchery (Lamar, Pennsylvania), an ozone concentration of 2-4 mg/L must be transferred to maintain a 0.2 mg/L ozone residual concentration after 10 minutes (Steven Summerfelt, personal communication). Cryer (1992) reported similar ozone demand results in tests on surface water supplies that were being disinfected at US Fish and Wildlife Service salmonid hatcheries in North America. All of the surface water supplies examined in these studies exhibit relatively high water quality with low concentrations of oxidizable organic material, iron, and manganese (Cryer, 1992; Steve Summerfelt, personal communication), yet the ozone demand created still reduced the half-life of ozone to less than a few minutes. In comparison, the half-life of ozone dissolved in pure water at 20°C is 165 minutes (Rice et al. 1981).

The ozone demand of water within RAS, which contains much higher levels of organic material and nitrite, creates a short ozone half-life, e.g., less 15 seconds, and makes maintaining an ozone residual difficult (Bullock et al. 1997). For this reason, it is difficult to add enough ozone to achieve microbial inactivation in recirculating systems. In recirculation systems ozone is most often applied at doses that promote water quality improvement (Brazil, 1996; Bullock et al. 1997; Summerfelt and Hochheimer 1997; Summerfelt et al. 1997). Using ozone in recirculating systems can reduce fish disease simply by improving water quality, which reduces or eliminates environmental sources of

stress (Bullock et al. 1997). These studies, as well as experience with ozone application at numerous commercial recirculating systems, indicates that both water quality and fish health can be improved by adding approximately 13–24 g ozone for every 1.0 kilogram of feed fed to a recirculating system.

"Rule of Thumb"

13–24 g ozone
per kilogram of feed.

Ozonation may enhance fine solids removal by changing particle size rather than separating particles from water. As an unstable reactive gas, ozone splits large organics into smaller biodegradable materials that can be more easily removed by heterotrophic bacteria. Conversely, ozone can polymerize metastable organics leading to enmeshment, direct precipitation, bridging, or adsorption (Reckhow et al. 1986). Ozone has been used sometimes with mixed success in a variety of aquaculture systems to remove color and turbidity (Colberg and Lingg, 1978; Williams et al. 1982; Paller and Lewis, 1988; Brazil, 1996; Summerfelt et al. 1997). Effects of ozonation upon particle size change in recirculating systems are still not clearly defined. The function of ozone is complicated; both qualitative and quantitative impacts of ozonation may be specific to a given system (Grasso and Weber, 1988). There are also concerns that even low ozone residuals may cause gill adhesions and mortality in fish exposed to freshly ozonated water (Rosenlund, 1975).

Besides being a method for water improvement in recirculation systems, ozone is valued for its high virucidal activity, Table 11.1. Among fish pathogenic viruses, high sensitivity toward ozone has generally been reported. This also applies to viruses with high UV resistance, i.e., IPNV and WSBV (Liltved et al. 1995; Chang et al. 1998). As viral diseases have become a major threat in worldwide aquaculture, the virucidal properties of ozone will certainly be more valued in the future, both in intake and effluent water disinfection. Ozone-treated water has also proven useful for washing fertilized eggs (Arimoto et al. 1996) and for reducing or eliminating potential pathogens associated with live prey such as rotifers in marine larval production systems (Davis and Arnold, 1997; Theisen et al. 1998).

OZONE DESTRUCTION

Creating an adequate level of residual ozone at the end of the contact chamber to ensure kill-off of bacteria will also necessitate that this same

ozone be removed prior to the water reaching the aquatic organisms. Ozone residuals can be lethal to fish at ozone concentrations as low as 0.01 mg/L, but the actual concentration depends upon species and life stage, Table 11.2. Due to the acute toxicity of residual ozone to aquatic animals (Wedemeyer et al. 1979), a de-ozonation unit has to be included. In many cases, residuals are eliminated by water retention within tanks immediately after ozonation, Fig. 11.5, or by applying small doses of a reducing agent, e.g., sodium thiosulphate. Dissolved ozone can also be stripped into air when passed through a forced-ventilation packed aeration column, Fig. 11.5. Air stripping will also remove dissolved oxygen concentrations that are in excess of saturation, which may or may not be desirable. Dissolved ozone can also be destroyed by passing the water through a biofilter or bed activated carbon, reaction with low levels of hydrogen peroxide, or contact with high intensity UV-light (catalyzing the conversion of O_3 to O_2).

Table 11.2 Toxicity of Dissolved Ozone to Fish (from Summerfelt and Hochheimer, 1997)

Species	Ozone concentration mg/L	Effect
Rainbow Trout	0.0093	96-h LC_{50}[a]
Rainbow Trout	0.01-0.06	lethal
Bluegill	0.01	60% mortality after 4 weeks
Fathead minnow	0.2-0.3	lethal
White perch	0.38	24-h LC_{50}
Bluegill	0.06	24-h LC_{50}
Striped bass (larvae)	0.08	96-h LC_{50}

[a]where LC_{50} is the concentration lethal to 50% of sa ple fish.

Achieving ozone destruction with UV electromagnetic radiation depends on the wavelength of the UV light source and the quantity of energy transmitted (Rodriguez and Gagnon, 1991; Hunter et al. 1998). Ozone residuals are destroyed at UV light wavelengths ranging from 250 to 260 nm. Ironically, UV wavelengths of 185 nm can be used to generate ozone. Ozonation by-products and their toxicity towards aquatic animals are not well described, especially in seawater. More long-lived reaction products are formed when brackish and seawaters are ozonated. Here, ozone reacts with bromide ions, and to a lesser extend with chloride ions, to form oxidants toxic to fish and shellfish. The most important are hypobromous acid (HOBr) and hypobromite ion (OBr-). Both have strong biocidal effects. By prolonged ozonation hypobromite ion can be further oxidized to bromate (BrO_3-), which is a persistent compound. In addition, small amounts of halogenated organic compounds like bromoform will be formed. Activated carbon filtration has been successfully used for removal of residual ozone and other oxidants in ozonated seawater (Ozawa et al. 1991).

RESIDUAL OZONE

Residual ozone in fresh water can be measured by the indigo colorimetric method, which is based on the decolorization of indigo by ozone (American Public Health Association, 1989). The decrease in light absorbance is linear with increasing concentration, and can be measured by a spectrophotometer at a wavelength of 600 nm. Because of the rapid reaction of ozone with bromide in seawater, it is not possible to measure dissolved ozone directly. Methods detecting the total residual oxidants (TRO) formed by ozone have been applied, including the DPD (N, N-diethyl-p-phenylenediamine) method and the iodometric method. As a surrogate parameter, the redox-potential is used for monitoring the oxidizing power of ozone and its oxidants in many practical applications. However, the absence of a linear relationship between residual ozone concentration and redox-potential is a disadvantage of this method.

MATERIAL RESISTANCE

Ozone is an extremely corrosive material when used in any water application. Materials that ozonated water will come in contact with must be selected with appropriate resistance properties. Table 11.3 provides a summary of materials and their corrosive resistance properties.

Table 11.3 Materials Resistance to Ozone Corrosion. (Adapted from Damez, F., 1982. Materials Resistant to Corrosion and Degradation in Contact with Ozone. In *Ozonation Manual for Water and Wastewater Treatement*. W.J. Masschelein (Ed.), JohnWiley & Sons, Ltd., NY NY.)

Materials	Ozonation Type of Exposure			Comments
	Dry air	Moist Air	Water	
METALS				
Chromium-nickel-silver; Brass	B*	B	B	
Aluminum & Al alloys	A	D	D	
Pig iron	A	A	A	Slow corrosion
Galvanized steel	B	C	C	Not shock resistant
Stainless steel	A	A	A	Without chlorine present
Sintered stainless steel	D	D	D	
PLASTICS[2]*& RUBBERS				
Kynar™, Viton™, Teflon™	A	A	A	Author's recommendation
Vinyl ester resin	A	A	A	
Polytetrafluoroethylene (Teflon ™)	A	A	A	
Polyamide (nylon-risen)	–	–	A	
Epoxide (Araldite™)	–	D	D	
Chlorosulfonated polyethylene (Hypalon™)	A	A	A	With appropriate charge
FPM (viton)	B	C	C	With appropriate charge
Silicone	D	C	C	With appropriate charge
Ethylene propylene	A	A	A	With appropriate charge
Polychloroprene (neoprene)	B	C	C	With appropriate charge
OTHER MATERIALS				
Concrete	A	A	A	
Glass & ceramics	A	A	A	
Fiberglass	Generally, epoxy vinyl ester resins are good with ozone and isophthalic resins are not; check with manufacturer.			

*NOTE: A = long lasting; B = usable; C = for low ozone concentration; and D = quickly degraded

[2] Author Note: PVC & CPVC are inadequate to carry pressurized ozone gas. PVC & CPVC are adequate for use in a continuously wetted gas-water transfer unit, hydraulic retention vessel, or when venting low-concentration wet off-gas from the transfer unit.

11.3 Factors Influencing Disinfection Efficiency

A variety of factors have been reported to influence the efficiency of bacterial inactivation by water and wastewater disinfection (Kostenbauder, 1991; Bessems, 1998; Heinzel, 1998). These include type of microbe, growth conditions of the microbe, type, and concentration of disinfectant, ability of the microbe to repair damages induced by the disinfectant, and water quality.

UV Repair Mechanisms

Bacteria may repair and reverse the lethal effects of UV irradiation by two major mechanisms: namely photoreactivation, and dark repair or liquid holding recovery. Both have the potential to increase the post-irradiation culturability by several $\log_{10}$ units as compared with the outcome of immediate plating and dark incubation. The combination of dark repair and photoreactivation generally leads to higher survival than dark repair alone, demonstrating that photoreactivation can revert lesions, which usually are not repaired in the dark (Moss and Davies, 1974).

Photoreactivation

The mechanism of photoreactivation requires visible light as a cofactor. Light in the near UV and visible spectrum (330-480 nm) activates DNA photolyase which reverts certain types of UV-induced damage, i.e., the UV-C and UV-B induced covalent bonds between adjacent thymines, without excising the distorted region (Jagger, 1967; Walker, 1984; Friedberg et al. 1995). The phenomenon was first recognized in the late 1940s when A. Kelner discovered the reversibility of UV damage on the spores of *Streptomyces griseus* by subsequent treatment with visible light (Kelner, 1961).

Conventional Michaelis-Menten enzyme kinetics has been used to describe the process of photoreactivation:

$$E + S \underset{k_2}{\overset{k_1}{\leftrightarrow}} ES \xrightarrow{k_3} E + P \tag{11.1}$$

In Eq. 11.1, E is the photoreactivating enzyme, S is the substrate (the photo-repairable DNA lesion), ES is the enzyme-substrate complex, and P is the repaired DNA lesion. The enzyme binds to the pyrimidine

dimers, and monomerises the dimers upon exposure to appropriate light. The reversible ES complex formation is light independent, while the k_3 is zero in the dark.

The rate of photoreactivation is strongly correlated with light intensity. Several hours are normally required for complete reactivation at lamplight intensities, while sunlight intensities have been reported to reduce the time for photoreactivation completion down to fractions of an hour in irradiated *E. coli* and halophilic bacteria (Harm, 1968; Eker et al. 1991). The photorepair mechanism is a widespread, but not universal trait among microorganisms, and there is no clearly defined phylogenetic distinction between the species, which have this ability, and those that have not. Viruses generally do not have the ability to photorepair. However, if the host cell has the required enzymes, photoreactivation of viruses may take place (Asato, 1976).

Dark Repair

Bacteria may use at least three different dark repair, or liquid holding recovery mechanisms: 1) nucleotide-excision repair, 2) SOS-error prone repair, and 3) post replication recombinational repair (Friedberg et al. 1995; Miller et al. 1999). A complex cellular mechanism, named the SOS regulatory system, is involved in all three dark repair processes.

The accurate nucleotide-excision repair process takes advantage of the fact that the information in DNA is present in two copies as a consequence of the complementary double-stranded nature of DNA (Swenson, 1976; Friedberg et al. 1995). An incision is introduced into the damaged strand near the site of the lesion, a DNA fragment which includes the damage is excised, and the missing DNA is then resynthesized by using the opposite strand as a template. In *E. coli*, dark repair is obtained by keeping the bacterium in nutrient-free buffer in the dark for several hours between UV irradiation and plating.

Dark repair processes have been found in some bacteria, including *E. coli* and moderate halophilic bacteria, and it has been proven absent from extreme halophilic Archaea (Fitt et al. 1983; Eker et al. 1991).

Water Quality

Particles

From drinking water and domestic wastewater investigations, it is reported that particles may provide protection against chemical and non-chemical disinfection agents, depending on the particle type and size, and the nature of association between the microorganism and the particle. Increased survival of particle-attached bacteria upon exposure to oxidants has mainly been attributed to: 1) high disinfectant reactivity at the particle surface, and 2) limited mass transfer at the interface. Each of these two factors will lead to a low concentration of oxidant available for inactivation of attached bacterial cells. However, this effect is highly dependent on the chemical demand of the particles. Failure of ozone to reduce the heterotrophic bacterial count more than one $\log_{10}$ unit in a recirculating rainbow trout culture system was attributed to rapid loss of ozone caused by elevated levels of total suspended solids (Bullock et al. 1997).

Several investigators have reported a correlation between suspended solids content and survival of faecal coliforms in UV-irradiated wastewater (Whitby and Palmateer, 1993). Qualls et al. (1983) observed that bacteria harbored by particles were partially protected so that UV disinfection was limited to a 3 to 4 $\log_{10}$ unit reduction in viability. Filtration was required to meet strict bacterial standards.

Scattering and absorption of UV light by particle surfaces may reduce the effect of radiation. Particles of organic origin will absorb more irradiation than mineral particles. Little protection of microbes is provided by clay particles because most of the irradiation is scattered. Shading may limit the exposure of individual bacteria, but has not been a problem in well-designed UV disinfection reactors with lateral dispersion.

In natural fresh and seawater sources, the particle content may rise in periods of high primary production or extreme weather conditions (wind and precipitation). In such periods, the inactivation process may be controlled by two different mechanisms: a rapid inactivation of single cells and cells associated with small particles, followed by a slower decay rate due to the partial protection of microbes associated with larger particles. Effective prefiltration and elevated disinfectant doses may sustain disinfection performance in such periods.

Dissolved Organic Matter

Dissolved organic carbon (DOC) is, by convention, defined as total organic carbon (TOC) after filtration through a 0.45 μm membrane filter. In freshwater supplies, humic substances originating from the terrestrial environment are often the most significant contributor to the DOC, conferring a brownish-yellow color on the water. In wastewater, proteins, carbohydrates, lipids, and amines will elevate the concentration of DOC. Oxidizing disinfectants like ozone will lose bactericidal strength through reaction with organic matter. The reaction products will generally have weak or no bactericidal activity. Hoigne (1988) has shown that aqueous ozone reactivity can be ascribed to two mechanisms: direct reactions

involving molecular ozone and reactions of active hydroxyl-radical intermediates produced by ozone decomposition.

Humic substances of natural waters are relatively resistant to ozonation. Sufficient contact time will produce small amounts of acetic, oxalic, formic and terephthalic acids, phenolic compounds and carbon dioxide. Generally, ozonated organic matter is more biodegradable than the original compounds. The instantaneous ozone demand of surface waters with DOC content of 2.5–3.5 mg/L has been reported to be in the range of 0.50–0.75 mg/L (Roustan et al. 1998). Surface water ozone demands of 0.4–0.5 mg ozone/mg DOC after 5 min of exposure at pH 7.5 was found by Graham (1999). The ozonolysis of carbon-carbon double bonds in organic molecules is an example of an ozone-demanding reaction.

> **"Rule of Thumb"**
>
> For UV applications, pre-filter water with a 50 micron screen.

Several organic compounds in water, e.g., humic substances, phenols and lignin sulfonates, will absorb UV radiation, thus reducing the doses available for microbial inactivation (Harris et al. 1987). The UV transmission of good quality seawater is generally high. However, the transmission may drop temporarily during bad weather conditions, in periods of planktonic blooms, etc. In general, assume you must use a 50 micron pre-filter to remove most of the particulate matter before applying UV. In shellfish operations, turbidity should not be above 20 nephelometric turbidity units (NTUs).

INORGANIC COMPOUNDS

Oxidizing disinfectants will react with inorganic compounds in accordance with their oxidation potential. Ozone will be involved in several redox reactions due to its high oxidation potential. Metal and heavy metal ions are oxidized to form stable compounds with low solubility. Ferrous and manganeous ions will react to ferric and manganic ions, respectively, which in turn will react with OH^- to form an insoluble precipitate. Bromide will be oxidized to bromate through several intermediary steps, while the reaction with chloride is limited by poor kinetics. The conversion of ammonia to nitrite is a slow, pH-dependent, first order reaction, while the nitrite is rapidly oxidized to nitrate. The latter reaction may have a significant effect on ozone disinfection capacity in wastewater treatment systems with incomplete

nitrification. Venosa (1983) reports that as much as 2 mg/L ozone was required to oxidize 1 mg/L of nitrite-N.

PH

Extreme pH values may inactivate microorganisms, or limit their growth. The activity of many disinfectants depends on pH. Small changes in the hydrogen ion concentration may influence the disinfection performance. The pH dependence of the biocidal activity of ozone is not clear. Reduced effect at high pH towards poliovirus and rotavirus as well as the cysts of the parasite *Naegleria gruberi* has been observed (Vaughn et al. 1987; Wickramanayake et al. 1991). However, the opposite relation was evident for *Giardia muris* cysts, which were more sensitive at pH 9 than at pH 5 and 7 (Wickramanayake et al. 1991). Changes in pH have little to no impact upon UV effectiveness.

TEMPERATURE

In general, the microbial inactivation rate by chemical disinfectants will increase with increasing temperature. Farooq et al. (1977) found a higher degree of ozone inactivation of *Mycobacterium fortuitum* at elevated temperatures. On the other hand, UV inactivation seems relatively insensitive to temperature. Negligible effects were observed in the range 5–35°C when pure cultures of *E. coli*, *Candida parapsilosis* and bacterial virus f2 were exposed to UV light in batch reactors (Severine et al. 1983).

11.4 AQUACULTURE WASTEWATER CHARACTERIZATION

Physical and chemical characterization of the water to be disinfected is of primary importance in order to select the best-suited pretreatment and disinfection method and to determine dose requirements. In the following, various influents and effluents of hatcheries, smolt farms, and processing industry have been characterized in relation to disinfection. These waters have been divided into five types on the basis of origin, concentration range of selected quality parameters and the objectives of disinfection, Table 11.4.

High content of organic matter indicates problems associated with disinfection by oxidants and UV irradiation. More extensive pretreatment than screening is advisable for such waters. In general, total suspended solids (TSS) and dissolved organic carbon (DOC) are important

indicators of disinfection performance, and usually there is a close correlation between DOC and the disinfectant demand of the water. In effluents, a more commonly used parameter is chemical oxygen demand (COD), which includes both particulate and dissolved organic matter. COD measurements are not suited for high salinity water due to interference by anions, especially chloride.

11.5 Inactivation Of Fish Pathogens

UV Irradiation

Data on inactivation of fish pathogenic bacteria and viruses by UV irradiation are summarized in Tables 11.5 and 11.6. The values show that UV doses of 2–6 mWs cm^{-2} reduce the viable count of the studied species by 99.9% or more in laboratory batch experiments. However, under more realistic continuous flow experiments with particles present, considerably higher doses are required to obtain a high degree of inactivation (Bullock and Stuckey, 1977; Liltved and Cripps, 1999). Therefore, inactivation doses obtained in the laboratory should be used with caution in predicting dose requirements in actual situations.

Studies by Torgersen (1998) indicate that the ISA virus is susceptible to UV light. Loss of infectivity was demonstrated when infected tissue homogenate was subjected to doses of 4–10 mWs cm^{-2}. In contrast, the IPN virus, a non-occluded birnavirus responsible of infectious pancreas necrosis in Atlantic salmon, is UV resistant. A dose of 122 mWs cm^{-2} was required to obtain 99.9% reduction in virus titer in brackish water (Liltved et al. 1995). Doses in the same order of magnitude have been experienced in studies conducted by Japanese investigators (Sako and Sorimachi, 1985; Yoshimizu et al. 1986). In spite of the resistance of IPNV, UV irradiation has become the method of choice for disinfection of supply water in Norwegian smolt farms where the UV units are designed for bacterial inactivation. The IPNV will pass through these installations unaltered since the doses applied are far too low for inactivation. Due to the substantial losses from IPN outbreaks, preventive measures such as upgrading of existing UV units to IPNV inactivating capacity when using water from sources suspected to carry infective levels of IPNV should be considered. Such upgrading will require at least a 5-fold increase in UV dose compared with the present 25 mWs cm^{-2} requirement.

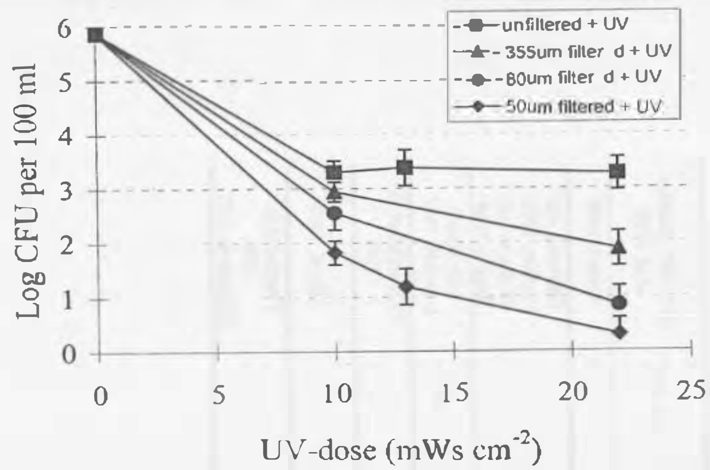

Figure 11.6 The effect of pre-filtration on survival of aerobic bacteria in UV irradiated water containing *Artemia* fragments (Liltved and Cripps, 1999).

Even higher doses have been required to reduce the infectivity of the Asian shrimp baculoviruses to zero. The mid-gut glad necrosis virus (BMNV) and the WSBV were inactivated by doses of 410 and 900 mWs cm^{-2}, respectively (Momayama, 1989; Chang et al. 1998). Due to the extreme resistance of these viruses and the excessive water flows required, UV irradiation is not a feasible method for baculovirus inactivation in Asian grow-out shrimp farms. However, high efficiency UV units could be suitable for hatchery and nursery ponds with limited water use. There are other studies providing additional information on the UV levels needed to achieve disinfection (Hunter et al. 1998; Wedemeyer, 1996).

Particle protection mechanism has been indicated for bacteria associated with Artemia fragments, due to lack of a dose dependent inactivation in the dose range of 10–22 mWs cm^{-2} (Fig. 11.6). The results obtained suggest a possible transmission of fish pathogenic bacteria to land-based aquacultural installations, even if the influent water is disinfected by UV irradiation. It was further demonstrated that prefiltration improved bacterial removal. Mesh sizes of 50 μm resulted in more than 5 $\log_{10}$ removal efficiency, indicating that influent water to aquacultural systems should be filtered to remove crustaceal fragments and other particles capable of harboring bacteria before UV disinfection.

Table 11.4 Water Parameters Affecting Disinfection Performance in Smolt Farms and Effluents From Salmon Filleting Industry & Slaughterhouses

Water type		TSS[a] mg/L	Turbidity NTU	COD mg/L	TOC mg/L	DOC mg/L	UV absorbance	Ozone demand mg/L	References
Influent water	Freshwater (good quality)	0.1–3.0	0.5–1	-	2.5–3.5	2.5–3.5	0.01–0.20	0.5–0.75	SFT 1997, Roustan et al. 1998
	Seawater	0.1–0.5	0.3–0.6	-	-	0.4-2	-	-	Valiela, 1984
Smolt farm effluent (normal tank operation)		0.2–15.3	-	6.0–50	0.3–4.3		-	-	Eikebrokk and Ulgenes, 1993; Bergheim and Åsgård, 1996
		22	3.8	-	9.8	6.6	0.100	3.9	Liltved and Landfald, 1995
Water in recirculating systems for salmonids		5.2–7.4	1.50–1.66	39.8–47.8	-	6.7–7.5	-	-	Summerfelt et al. 1997
Effluent from salmon filleting plant		1600	-	3050	-	-	2.27	-	Liltved, 1997
Effluent from salmon slaughterhouses		40–1375	-	1500–3000	567	-	1–20	-	Flugstad et al. 1991; Millamena, 1992

[a]Definitions = TSS total suspended solid
NTU nephleometric turbidity units
COD chemical oxygen demand
TOC total organic carbon
DOC dissolved organic carbon

Table 11.5 Inactivation of Pathogenic Microorganisms by UV Irradiation

Organism	Percent removal	UV dose (milli-Ws cm^{-2})	Temp. (°C)	pH	Water	Reactor type	Comments	References
Bacteria								
A. salmonicida *A. punctata* *A. hydrophila* V. anguillarum *P. fluorescens*	99.99	22				Continuous flow	No info about water quality	Kimura *et al.* 1976
A. salmonicida *A. hydrophila* V. anguillarum P. fluorescens *Y. ruckeri*	99.9-100	21-24	12.5		Spring water	Continuous flow	UV trans.: 95% 1cm Turbidity: 4JTU	Bullock and Stuckey 1977
A. salmonicida V. anguillarum V. ordalii	99.9	2.9-5.5				Batch	No info about water quality	Sako and Sorimachi 1985
A. salmonicida *V. anguillarum* *V. salmonicida* *Y. ruckeri*	99.999	2.7	Room temp.		Brackish	Batch	15 ‰ salinity	Liltved *et al.* 1995
A. salmonicida	99.9	2.4	7	7.2	PBS	Batch		Liltved and Landfald 1995
A. salmonicida	99.9	2.5	7	7.8	Fish farm effluent	Batch	TSS 22 mg/l TOC 9.8 mg/l	Liltved and Landfald 1995
A. salmonicida *V. anguillarum* *Y. ruckeri*	99.9	1.2-3.2	7	7.2	PBS	Batch		Liltved and Landfald 1996
A. salmonicida *V. anguillarum* Y. ruckeri	99.9	8.5-13.4	7	7.2	PBS	Batch	With photo-reactivation	Liltved and Landfald 1996
HPC	<99.9	22			Seawater	Continuous flow		Liltved and Cripps 1999

Organism	Percent removal	UV dose (mWs cm^{-2})	Temp. (°C)	pH	Water	Reactor type	Comments	References
Viruses								
IHNV	99.9	2				Batch		Sako and Sorimachi 1985
IHNV	99	1-3				Batch		Yoshimizu *et al.* 1986
IPNV	99.9	150-200				Batch		Sako and Sorimachi 1985
IPNV	99	100-150				Batch		Yoshimizu *et al.* 1986
IPNV	99,9	122	Room temp.		Brackish	Batch	15 ‰ salinity	Liltved *et al.* 1995
IPNV	99.9	246	5	7.9	Seawater	Batch	Natural	Liltved *et al.* 2006
AHNV	99.9	104	5	7.9	Seawater	Batch	Natural	Liltved *et al.* 2006
ISA-virus	99.9	7.5	5	7.9	Seawater	Batch	Natural	Liltved *et al.* 2006
ISA-virus	Loss of infectivity	4-10			Tissue homogenate	Batch		Torgersen 1998
WSBV	Loss of infectivity	900			Virus stock solution	Batch	20 ppt seawater	Chang *et al.* 1998

HPC - heterotrophic plate count
IHNV – infectious hematopoietic necrosis virus
AHNV - Atlantic halibut nodavirus
ISA – infectious salmon anaemia
WSBV – white spot syndrome baculovirus

Table 11.6 UV Energy for a 100% Kill at Wavelength of 2537 Angstrom) (from Philips and Hanel, 1960; also available from Wheaton, 1977)

Organism	UV Energy (micro-Watt Second per square centimeter, m Ws cm^{-2})
Mold Spores	
Penicillium roqueforti	26,400
Aspergillus glaucus	88,00
Aspergillus niger	330,000
Yeasts	
Brewer's yeast	6,600
Baker's yeast	8,800
Common yeast cake	13,200
Saccharomyces sp.	17,600
Bacteria	
Streptococcus hemolyticus	5,500
Staphylococcus aureus	6,600
Escherichia coli	7,000
Proteus vulgaris	7,500
Bacillus subtillis	11,000
Bacillus subtillis spores	22,000
Sarcina lutea	26,400
Virus	
Bacteriophage (*E. coli*)	6,600
Influenza virus	3,400
Nematode EGGS	40,000

Absence of toxic residues and reaction products, low costs when treating high quality water, easy operation and maintenance, and minimal space requirements, are main advantages of UV irradiation. Fouling of the quartz sleeves must be dealt with on a regular basis, normally by mechanical or chemical cleaning. The major disadvantage of this method in aquacultural applications is the inefficiency against important fish pathogenic viruses. This might limit the use of UV irradiation in the future as virus control becomes a more important preventive measure in the fish and shrimp farming industry.

PHOTOREACTIVATION

Photoreactivation and dark repair have not previously been studied in fish pathogenic bacteria. In the present study it was demonstrated that *A. salmonicida, V. anguillarum* and *Y. ruckeri* are able to repair UV damage both in the presence and absence of visible light (Liltved and Landfald, 1996; Liltved and Landfald, 2000). Photoreactivation improved survival of these fish pathogens to about the same degree as has been found for common water quality indicator bacteria under similar experimental conditions (Harris et al. 1987).

At a light intensity typical of indoor aquacultural facilities (1500 lx), 10- to 1000-fold recoveries were obtained within the first two hours in *A. salmonicida, V. anguillarum* and *Y. ruckeri* (Liltved and Landfald, 1996). Photoreactivation was essentially completed within 4–6 hours. These trials were conducted in the laboratory with bacterial cells suspended in buffers.

UV irradiation followed by high-intensity light repair was also studied under more realistic conditions. UV exposed *A. salmonicida* and *V. anguillarum* suspended in their natural water qualities, respectively, demonstrated a less pronounced, but more rapid photoreactivation at sunlight intensities than in lamplight. Only 20 minutes and 1 hour respectively was required for complete reactivation (Liltved and Landfald, 2000). These trials show that even in indoor facilities with elevated light fluxes, water retention times, and/or temperatures, recovery processes may elevate the number of viable bacteria after irradiation. More rapid reactivation may occur in UV disinfected supply water and wastewater if exposed to sunlight in outdoor tanks or water recipients.

As compared to drinking water supply systems, the influence of photoreactivation is likely to be more pronounced in aquaculture facilities. In potable water systems, UV disinfection is usually the last step in a succession of treatments. Between this point and the end user there is normally no photoreactivating light available. UV irradiated water in aquacultural facilities will be exposed to artificial light or sunlight (daylight) capable of initiating photoreactivation.

Significant light-induced recoveries should not be neglected, and should be taken into consideration when assessing the efficiency of UV disinfection of aquacultural water. As a rule, the applied UV dose should account for the worst case with respect to reactivation. In Norwegian smolt farms, the veterinary authorities have demanded a minimum UV dose of 25 mWs cm^{-2}. The practical experience with this dose from about 100 UV installations has been good, with only a few outbreaks of bacterial diseases suspected to be transmitted through UV disinfected influent water.

An UV dose of 25 mWs cm^{-2} should be sufficient to suppress detectable repair of the fish pathogenic bacteria studied. However, if the dose is reduced for some reason, i.e., due to reduced lamp intensity caused by aging or fouling of the quarts sleeves, reduced water quality, increased water flow, or partly protected bacteria associated with particles, photoreactivation might elevate the number of survivors to infective levels.

DARK REPAIR

Dark repair, or liquid holding recovery, in fish pathogenic bacteria (*A. salmonicida, V. anguillarum* and *Y. ruckeri*) was found to be less efficient and slower than photoreactivation (Liltved and Landfald, 1996). Even at an elevated temperature of 22°C, 48 hours or more was required for completion. In aquacultural production units, like salmon hatcheries, which are characterized by relatively cold water and short hydraulic retention times, liquid holding recovery alone cannot be expected to have strong impact on post-UV irradiation survivability within the production units. Continuation of the recovery process is probable after disposal of effluent water to the recipient. This could significantly influence estimates of environmental impacts of release of UV treated aquacultural water. Dose-survival curves of UV-irradiated *A. salmonicida* after various post-irradiation treatments for different dark liquid holding periods and illumination levels are shown in Fig. 11.7 (Liltved and Landfald, 1996).

11.6 OTHER METHODS OF DISINFECTION

There are of course many other means of disinfecting water, equipment, and instruments used in an aquaculture setting. Each has its proponents and each has its understood advantages and disadvantages. Some brief recommendations on chlorine dosages and contact times for disinfecting various pieces of equipment are given in Table 11.7.

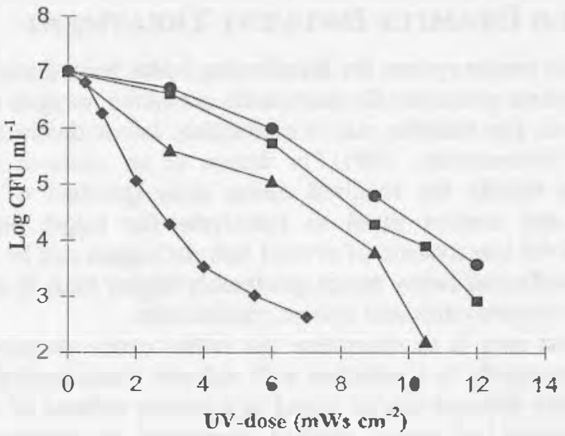

Figure 11.7 Dose-survival curves of UV-irradiated *A. salmonicida* after various post-irradiation treatments. All incubations were done at 22°C. Symbols: ♦, immediate plating/dark incubation; ▲, dark liquid holding for 48 h; ■, illumination at 1500 lx for 8 h; •, illumination followed by dark holding in nutrient free buffer (Liltved and Landfald, 1996).

Table 11.7 Recommendations on Chlorine Dosages & Contact Times (Lawson, 1995)

Equipment Description	Contact Time Minutes	Chlorine Form: Bleach (mg/L)	Chlorine Form: Calcium Hypochlorite (mg/L)
Nets, buckets boots, etc	5	0.70	40
Transport equipment	30	2.64	150
Rearing containers	60	3.51	285

Table 11.8 compares the relative oxidation potential of several known oxidants (Lawson, 1995). Table 11.9 provides an overall summary of common disinfectants.

Table 11.8 Relative Oxidation Potentials of Known Oxidants (Lawson, 1995)

Material	Volts	Oxidation Potential
Fluorine	2.87	Most Reactive
Ozone (O_3)	2.07	↓
Hydrogen Peroxide (H_2O_2)	1.78	
Potassium permanganate ($KmnO_4$)	1.70	
Hypobromus acid (Hobr)	1.59	
Hypochlorous acid (HOCl)	1.49	
Chlorine (Cl_2)	1.36	
Chlorine dioxide (ClO_2)	1.27	
Oxygen (O_2)	1.23	
Bromine (Br_2)	1.09	
Iodine (I_2)	0.54	Least reactive

OSHA PERMISSABLE LIMITS FOR OZONE in AIRSPACES

Time weighted average (TWA):	0.1 ppm
Short-term exposure limit (STEL):	0.3 ppm
Immediately Dangerous to Life or Health (IDLH):	5.0 ppm

More information at the OSHA website:
http://www.osha.gov/dts/chemicalsampling/data/CH_259300.html

Table 11.9 Comparison of Actual and Ideal Characteristics of Common Disinfectants

Characteristic	Property/ Response	Chlorine	Sodium Hypochlorite	Calcium Hypochlorite	Ozone	UV Radiation
Availability	Should be available in large quantities and reasonably priced	Low cost	Moderately low cost	Moderately low cost	Moderately high cost	Moderately high cost
Interaction with extraneous material	Should not be absorbed by organic matter other than bacterial cells	Oxidizes organic matter, also absorbed by organics	Active oxidizer	Active oxidizer	Oxidizes organic matter	Absorbed by specific organic compounds
Noncorrosive and nonstaining	Should not disfigure metals or stain clothing	Highly corrosive	Corrosive	Corrosive	Highly corrosive	Not Applicable
Nontoxic to higher forms of life	Should be toxic to microorganisms and nontoxic to humans and other animals	Highly toxic to higher life forms	Toxic	Toxic	Toxic	Toxic at high dosages
Penetration	Should have capacity to penetrate surfaces (bacteria)	High	High	High	High	High
Safety	Should be safe to transport, store, handle and use	High risk	Moderate risk	Moderate risk	Moderate risk	Low risk
Solubility	Must be soluble in water or cell tissue	Slight	High	High	Low	Not applicable
Stability	Loss of germicidal action on standing should be low	Stable	Slightly unstable	Relatively stable	Unstable, must be generated as used	Must be generated as used
Toxicity to microorganism	Should be highly toxic at high dilutions	High	High	High	High	High
Toxicity at ambient temperatures	Should be effective in ambient temperature range	High	High	High	High	High

Source: Water Environment Research Foundation

11.7 Design Example Influent Treatment

Design of an ozone system for disinfecting water immediately before fish culture requires processes for particulate exclusion, oxygen delivery, ozone generation, gas transfer, ozone contacting, ozone destruction, and gas balancing (Summerfelt, 2003).For design of an ozone-system, the first step is to decide the required ozone dose (product of residual concentration and contact time) to inactivate the target organisms. Required doses for inactivation of several fish pathogens can be found in Table 11.1. A sufficient safety factor (preferably higher than 2) should be used to include uncertainties and system variabilities.

The next step is to determine the initial ozone demand of the actual water, preferably in a situation with reduced water quality ("worst case"). The ozone demand can be found in a known volume of water by direct measurement of ozone applied compared to residual ozone concentration measured. Alternatively, the ozone demand can be estimated based on the content of ozone demanding substances. Sand-filtered water low in suspended solid with a maximum dissolved organic carbon (DOC) content of 5 mg/L, corresponding to an initial ozone demand of approximately 2 mg/L (see Section 11.3). The sum of the ozone demand and residual ozone concentration will be the required amount of ozone applied to the water.

Also for design of UV-systems, the first step is to decide the required UV-dose (product of UV-intensity and retention time in the UV-chamber) to inactivate the target organisms, see Tables 11.5 and 11.6. It is important to use high enough safety factors. One special feature to consider is the ability of fish pathogenic bacteria to repair DNA-damage induced by UV-irradiation. To compensate for repair, a factor of 4 should be used. In addition, a factor of at least 2 should be used to include other uncertainties and system variabilities. Particles should be removed before UV-disinfection.

The next step is to measure the UV-transmission or the UV-absorbance of the actual water under "worst case" conditions. These measurements say something about how much of the UV-light that will penetrate a specified path length (normally 5 cm) of the water, and are essential parameters for proper design. Most dealers of UV-systems will have good models to use for design of UV-units based on UV-transmission/absorbance and UV dose.

Design Example: U.S. Fish and Wildlife Service's Northeast Fishery Center in Lamar, Pennsylvania

You will extract 0.5 m^3/s of flow from the flowing stream. Design a UV/Ozone system to treat the incoming water.

Solution

This example is based on the water filtration and ozone disinfection system installed at the U.S. Fish and Wildlife Service's Northeast Fishery Center in Lamar, Pennsylvania, which treats a surface water supply that is used to culture endangered fish as described by Summerfelt et al. (In Press). In bench-top studies, the ozone demand required to maintain a 0.2 mg/L residual after 10 minutes was determined to be 2.5 mg/L. To overcome the ozone demand of the water, the ozonation system was sized large enough to dose a maximum ozone concentration of approximately 5 mg/L to a design flow of 1,500 L/min.

The treatment system first passes the surface water through two parallel drum filters operated with 60-µm sieve panels in order to exclude the majority of debris, algae, and organisms larger than the sieve openings. After microscreen filtration, two or three variable speed pumps are operated in parallel to supply between 400 to 2,400 L/min to the ozone treatment system. Two corona discharge type ozone generators, each with a remote controlled 20:1 turndown capacity, were installed to provide redundant, backup capacity. Ozone contained within an approximately 95% oxygen feed gas exits the generator and is transferred in to the water (at 0.5-0.7 bar) through a down flow bubble contactor following each pump.

The ozonated water is then piped to a 15 m^3 ozone contact column. The contact column provides approximately 20, 10, or 6.7 minutes of plug-flow contact time for water flows of 760, 1,500, or 2,270 L/min, respectively. A dissolved ozone probe at the outlet of the ozone contact chamber continuously monitors the dissolved ozone concentration discharged from the contact tank. A proportional-integral-derivative feed-back control loop is used to adjust the concentration of ozone generated (and thus added) in order to maintain the dissolved ozone residual discharged from the ozone disinfecting contact tank at a pre-selected set-point (nominally 0.2 mg/L). This application provides a relatively high ozone dose and contact time, i.e., 0,2 mg/L·min of dissolved ozone residual remaining after the water flow exits the disinfecting contact tank.

The water discharged from the ozone disinfecting contact tank then flows by gravity through a second 32 m^3 contact tank, which provides additional time for the dissolved ozone to decompose. Any dissolved

ozone remaining in the water exiting the second contact vessel is air-stripped, along with any large dissolved oxygen super-saturation, as the water flows by gravity through a forced-ventilated cascade column. This treated water then flows by gravity to the fish culture systems. Summerfelt et al. (In Press) provide a more detailed description of this case study design example.

The following equations were required to determine the mean hydraulic retention times, the mass ozone application rate, the ozone dose applied, and the resulting ozone disinfecting C*t using the water flow rate, the ozonated gas supply flow rate, the ozone gas concentration, and the dissolved ozone concentration following the disinfection contact tank as inputs. The variables in square brackets are provided by system specifics:

where Q_{water} is treatment flow rate
$V_{contact\ tank}$ is contact tank volume
Q_{oxygen} is oxygen gas flow rate
Ozone residual concentration is desired concentration of O_3 in water leaving contact tank

$$\text{Contact time} = \left\{\frac{1}{[Q_{water}]}\frac{\text{min}}{\text{L}}\right\}\left\{\frac{3.78\text{L}}{\text{gal}}\right\}\left\{[V_{contact\ tank}\ \text{gal}]\right\} = \#\#\ \text{min}$$

$$\text{Mass}_{applied\ Ozone} = \left\{[Q_{oxygen}]\,{}^{\text{L}}\!/_{\text{min}}\right\}\left\{{}^{\text{m}^3}\!/_{1000\text{L}}\right\}\left\{\frac{1.331\text{kgO}_2}{\text{m}^3}\right\}\left\{\frac{1440\text{min}}{\text{day}}\right\}$$
$$\left\{\frac{\text{molO}_2}{32\text{gO}_2}\right\}\left\{\frac{2\text{molO}_3}{3\text{molO}_2}\right\}\left\{\frac{48\text{gO}_3}{\text{molO}_3}\right\}\left\{\frac{[\text{O}_3\text{percentage in feedgas}]}{100}\right\}$$
$$= \#\#\ {}^{\text{kg O}_3}\!/_{\text{day}}$$

$$\text{Concentration}_{applied\ Ozone} = \left\{[\text{Mass}_{applied\ Ozone}]\frac{\text{kgO}_3}{\text{day}}\right\}\left\{\frac{10^6\text{mg}}{\text{kg}}\right\}\left\{\frac{1}{[Q_{water}]}\frac{\text{min}}{\text{L}}\right\}\left\{\frac{\text{day}}{1440\text{min}}\right\}$$
$$= \#\#\ {}^{\text{mg}}\!/_{\text{L}}\ \text{of Ozone Applied}$$

$$\text{Ozone Disinfecting C}\cdot\text{t} = \left\{\text{Ozone residual concentration}\ {}^{\text{mg O}_3}\!/_{\text{L}}\right\}\left\{\text{Contact time, min}\right\}$$
$$= \#\#\ {}^{\text{mg O}_3}\!/_{\text{L}}\cdot\text{min}$$

11.8 Conclusions

- Fish pathogenic bacteria are generally susceptible to UV irradiation. A 99.9% or higher inactivation has been obtained by doses of 1.5–3.4 mWs cm^{-2} in high quality natural waters and buffers. Contrasting this susceptibility of bacteria, some viruses (including the IPN-virus and Asian shrimp baculoviruses) are highly resistant to UV irradiation, demanding up to several hundreds mWs cm^{-2} for equivalent bacterial inactivation. This implies that UV units must be designed with high intensity irradiation and low water flow to obtain the required UV dose to inactivate these viruses.
- When bacteria associated with particles are UV-irradiated, protection has been demonstrated (Liltved and Cripps, 1999). Improved overall bacterial removal was demonstrated using particle prefilters with mesh sizes of 50, 80, and 355 µm. More than 5 $\log_{10}$ units total reduction in viable count was obtained with a 50-µm pre-filter prior to a UV dose of 22 mWs cm^{-2}, compared to less than 3 $\log_{10}$ reduction without filtration. Consequently, water for aquacultural purposes should be filtered to remove crustaceal fragments and other particles capable of harbouring bacteria before UV disinfection, thereby reducing the risk of introducing UV-shielded bacteria.
- It has been shown that fish pathogens, including the IPN-virus, are susceptible to ozone. A 99.9% or higher inactivation has been obtained at residual ozone concentrations of 0.15–0.20 mg/L within 60 s in natural lake-, brackish- and seawater. However, new results indicate that some viruses, including IPNV and AHNV, are highly resistant to ozonated seawater.The logarithmic inactivation curves are often characterised by a rapid inactivation initially, followed by a decreasing inactivation rate with exposure time. Such kinetics could be explained by reduced ozone concentration during the course of exposure. In a fish farm effluent, a residual concentration of 0.3–0.4 mg/L was required for *A. salmonicida* inactivation. In practical ozonation of fish farming water, it is important that the ozone dose is high enough to account for the initial demand, thereby establishing a sufficient residual concentration for the required contact time.
- The three fish pathogenic bacteria *A. salmonicida, V. anguillarum* and *Y. ruckeri* are all able to photo-reactivate and exhibit liquid holding recovery when subjected to suitable post-UV-C irradiation conditions. With these two reactivation mechanisms, alone or in combination, the 99.9% inactivation dose has to be raised 3–4 times to obtain the same inactivation as with normal bacterial enumeration procedures, i.e., plating and dark incubation.
- Photo-reactivation is the more efficient reactivation mechanism by causing both a more rapid and a more complete recovery than holding in the dark. Light intensity and temperature influenced the rate of photo-reactivation. From 10 to 1000-fold recoveries were obtained within the first two hours at light intensities and temperatures characteristic of indoor salmonid aquacultural facilities. At sunlight intensities, photo-reactivation was completed within 20 minutes in *A. salmonicida*, while 60 minutes was required in *V. anguillarum*. Photo-reactivation should be accounted for when assessing the necessary doses for efficient UV disinfection of aquacultural waters.
- An ozonation system is relatively complex and expensive compared to UV irradiation. The process becomes even more complex if oxygen is produced on-site.

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CHAPTER 12

FLUID MECHANICS AND PUMPS

12.0 FLUID MECHANICS

Fluid mechanics is the study of the impact that forces have on fluids, and it includes fluid statics (stationary fluids) and fluid dynamics (fluids in motion). This chapter is no substitute for formal course work and studies of fluid mechanics, but is intended to introduce the reader to some of the fluid mechanics principles that are most pertinent to aquaculture. The reader should consult a fluid mechanics text for more in depth treatment of the various topics.

In engineering terms, a fluid is a substance that deforms continuously when subjected to a shear force. In aquaculture, water and air are the two fluids of prime interest. Water is a liquid at ordinary temperatures and pressures, although it also exists in small quantities as a gas under these conditions. As a liquid, it can be treated as an incompressible fluid. Air, on the other hand, is a compressible gas. The two important physical properties of fluids are density and specific weight. Density is defined as mass per unit volume (kg/m^3), and specific weight is defined as weight per unit volume (lb/ft^3). The specific gravity of a fluid is the ratio of its density to the density of water, and is unitless.

The pressure of a fluid is defined as the normal force exerted over a unit area, and is usually expressed as pounds per square inch (psi or Pascals). For example, the pressure at the bottom of a column of water is equal to the specific weight of the fluid (N/m^3 or lb/ft^3) times the height of the fluid column (m or ft) above the point in question. Pressure measurements can also be referenced to either absolute zero or atmospheric pressure. If the pressure reading is referenced to absolute zero or a vacuum, the measurement is referred to as absolute pressure or psia. Thus, atmospheric pressure at mean sea level is given as 101 kN/m^2, 14.7 psia, 760 mm of mercury (Hg) or 101.3 kPa. Common pressure gauges measure the difference between the pressure of the fluid being measured and the pressure of the surrounding atmosphere or psig. For example, a tire gauge measures the air pressure in comparison to the existing barometric pressure. Thus, gauge pressure is equal to the difference in absolute pressure and actual atmospheric pressure. Since

gauge pressure is referenced to atmospheric pressure, it can be either positive or negative. Negative gauge pressure is also referred to as vacuum pressure.

Pressure is normally measured using either a manometer or by measuring the deformation of an elastic member, whose deflection is directly proportional to the applied pressure (tradition pressure gauges). A manometer uses the change in height of a column of liquid to measure pressure. In its simplest form, a manometer or piezometer is a straight vertical tube attached to a pipe. The gauge pressure at the center of the pipe is equal to the product of the specific weight of the fluid (N/m^3) times the height of the fluid column (cm or inch). In practice, a section of Tygon™ tubing or clear acrylic tubing attached to a threaded barbed pipe adapter makes a simple, flexible piezometer. These are often placed on the discharge pipe into a tank to monitor the backpressure and thus indirectly the flow into the tank (since flow is proportional to the square root of pressure).

The study of fluids in motion is fluid dynamics and is based on several fundamental concepts including conservation of mass, energy, and Newton's Laws of motion. The law of conservation of mass, which says that mass can neither be created nor destroyed, yields the concept of the continuity equation. For pipe flow, this means that the mass of fluid flowing past one section of a closed pipe must also flow past all other sections, since there is no where else for it to go. Using the continuity equation, the pipe velocities can be easily calculated knowing the flow rate through the pipe (Fig. 12.1):

$$Q = \rho_1 A_1 V_1 = \rho_2 A_2 V_2 \qquad (12.1^a)$$

In addition, the velocity through pipe sections of different diameters can be determined since the fluid density is the same, hence:

$$V_1 A_1 = V_2 A_2 \qquad (12.2)$$

$$A = \frac{\pi D^2}{4} \qquad (12.3)$$

The magnitude of the water velocity is a crucial variable, as it affects the pressure losses through pipe runs, turns, and fittings.

[a] Symbols are defined at end of chapter.

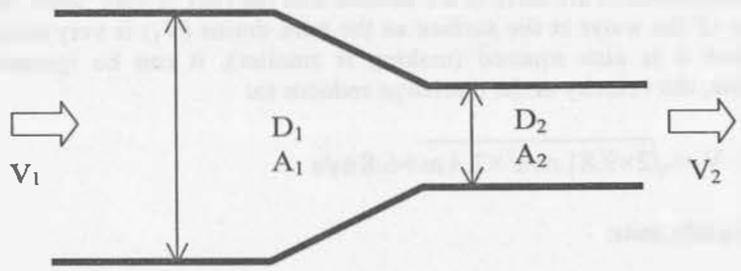


Figure 12.1 Pipe cross-section and pertinent terms.

Another important concept is the law of conservation of energy. The total energy at any point in a fluid consists of three components: potential energy due to location, i.e. height, potential energy due to pressure, and kinetic energy due to the motion of the fluid, i.e. velocity. Potential energy is due to elevation above some point of reference. For example, water in an overhead tank has potential energy in relationship to the pipe located at floor level. The potential energy is a function of the weight of the water and the vertical distance from the pipe to the water level in the overhead tank. An example of potential energy due to pressure is the energy (pressure) added by a pump. Finally, the kinetic energy due to the motion of the fluid is directly related to its velocity using Newton's laws of motion. The total energy of the fluid at some point is the sum of the energies due to elevation, pressure, and velocity.

From the law of conservation of energy, the total energy at any point in a fluid system must be the same. This yields Bernoulli's equation, which is usually expressed as:

$$E_1 = Z_1 + \frac{P_1}{\gamma} + \frac{V_1^2}{2g} = Z_2 + \frac{P_2}{\gamma} + \frac{V_2^2}{2g} = E_2 \qquad (12.4)$$

The term (P_1/γ) is often referred to as the pressure head and the term ($V^2/2g$) as the velocity head or dynamic head, since both can be expressed in feet (meters). As an example how Bernoulli's equation is used, calculate the velocity of the discharge from a storage reservoir tank that is 2.44 m (8 feet) deep. We know that the energy at the top of the tank and at the discharge point must be the same. In addition, since the

system is open to the atmosphere at the top and at the discharge point, the gauge pressures are zero. If we assume that the tank is very large, the velocity of the water at the surface as the tank drains (V_1) is very small and since it is also squared (making it smaller), it can be ignored. Therefore, the velocity at the discharge reduces to:

$$V_2=\sqrt{2\times 9.81\,m/s^2\times 2.4\,m}=6.8\,m/s$$

or in English units:

$$V_2=\sqrt{2\times 32.2\,ft/s^2\times 8\,ft}=22.7\,ft/s$$

Bernoulli's equation can thus be used to calculate the specific energy components at a particular point in the system and relate how one form of energy, potential or kinetic, is converted into another. A broader form of the conservation of energy law would take into account other forms of energy that might be added or removed from a system, including energy added by a pump, energy extracted via a turbine, or energy lost due to friction in pipes and fittings:

$$E_1+E_{added}=E_2+E_{lost}+E_{extracted} \tag{12.5}$$

The energy "lost" to friction is actually transformed into thermal energy, consistent with the concept of energy conservation mentioned above. Energy inputs to a system are normally included on the left side of Eq. 12.5 as a positive value. Energy losses due to pipe friction and minor losses in fittings are added to the right side of the equation, since this is energy lost between points 1 and 2. Pipe friction losses are caused by the friction generated by the movement of the fluid against the walls of the pipes. Minor pipe loses are those associated with friction occurring when the fluid encounters restriction in the systems (valves), changes in direction (elbows, bends, tees), changes in pipe size (reducers, expanders), and losses when the fluid enters or leaves a conduit. Sometimes minor losses are referred to as fitting losses; since "minor" can be a misnomer in that these pressure losses can be large.

12.1 FRICTIONAL LOSSES

The magnitude of pipe friction losses is a function of the internal pipe diameter, length, fluid velocity, roughness of the internal pipe surfaces and certain physical properties of the fluid such as density and viscosity. Over the years, there have been several empirical relations developed to relate these factors to the pipe friction head loss. One of these is the Darcy-Weisbach equation, which first requires the calculation of the Reynolds number (Re) to determine the type of flow, i.e. laminar or turbulent. Then the relative roughness coefficient and the Moody's diagram (see Fig. 12.2) are used to find the friction factor and finally the head loss using Eq.12.6. One of problems with this method is that all of the coefficients are estimated for average or normal conditions, without considering the effects of biofouling for example. Thus the choices of the coefficient values are at best an educated guess as to the current or projected conditions, which probably will and do change with time. The head losses resulting from various water flow rates in plastic pipe can be calculated by means of the Hazen-Williams formula as opposed to the Darcy-Weisbach equation. Both methods will be demonstrated in this chapter.

Darcy-Weisbach

$$H_L=f\left(\frac{L}{D}\right)\left(\frac{V^2}{2g}\right) \tag{12.6}$$

Hazen-Williams

$$H_L=0.2083\left(\frac{100}{C_{H\text{-}W}}\right)^{1.852}\left(\frac{Q^{1.852}}{D_{inch}^{4.87}}\right) \tag{12.7}$$

where H_L in the Hazen-Williams equation, Eq. 12.7, is per 100 ft of pipe and $C_{H\text{-}W}$ is 150 for PVC pipe and 100 for cast iron pipe.

The Reynolds number, Re, is a dimensionless number and is the ratio of inertial forces to frictional forces. Re numbers are calculated as the product of velocity and pipe diameter (inertial forces) divided by kinematic viscosity (viscous forces):

$$Re=\frac{VD}{\nu} \tag{12.8}$$

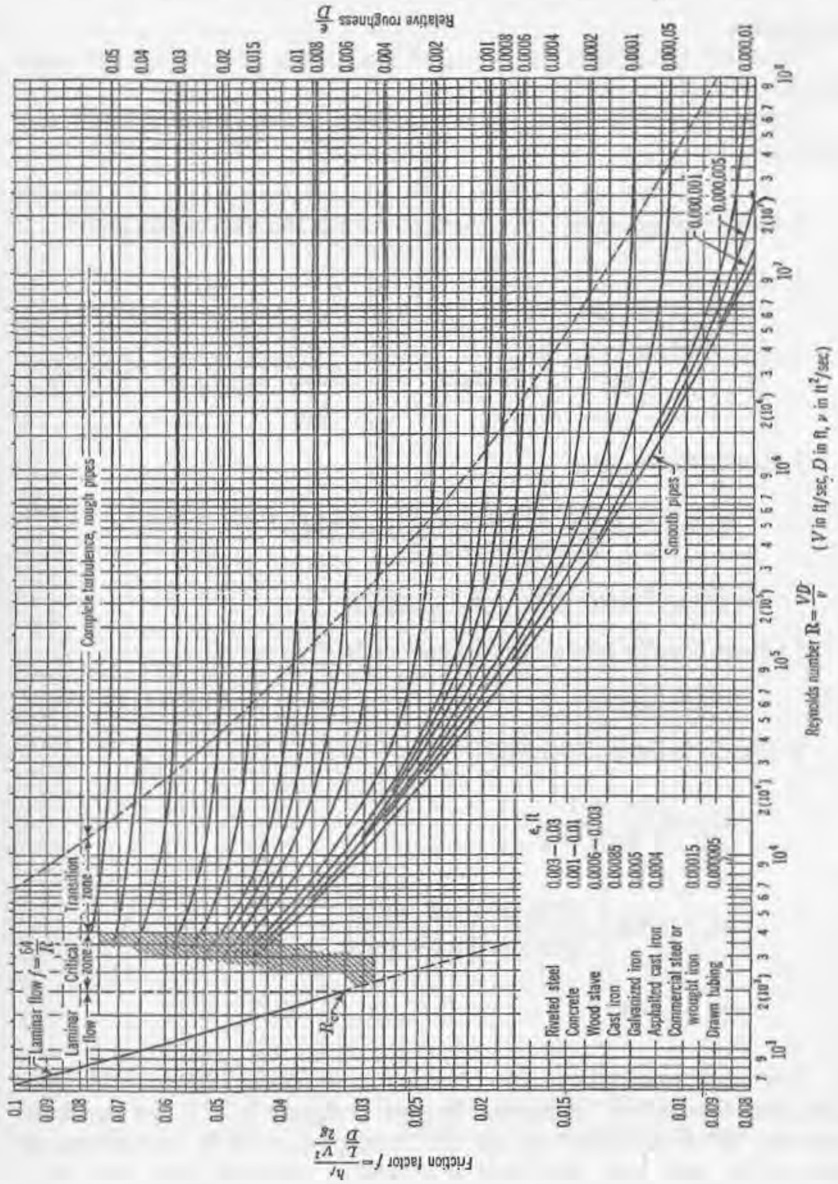


Figure 12.2 Moody diagram.

Table 12.2 Velocity, Dynamic Head and Reynolds Number

Velocity Look up Table in ft/s	Diameter of Pipe in Inches					
Flow, gpm	2	3	4	6	8	12
1	0.102	0.045	0.026	0.011	0.006	0.003
5	0.511	0.227	0.128	0.057	0.032	0.014
10	1.021	0.454	0.255	0.113	0.064	0.028
25	2.553	1.135	0.638	0.284	0.160	0.071
50	5.105	2.269	1.276	0.567	0.319	0.142
100	10.211	4.538	2.553	1.135	0.638	0.284
250	25.527	11.345	6.382	2.836	1.595	0.709
500	51.054	22.690	12.763	5.673	3.191	1.418
1,000	102.107	45.381	25.527	11.345	6.382	2.836
2,000	204.214	90.762	51.054	22.690	12.763	5.673
4,000	408.428	181.524	102.107	45.381	25.527	11.345

Dynamic Head Look up Table in ft	Diameter of Pipe in Inches					
Flow, gpm	2	3	4	6	8	12
1	1.62E-04	3.20E-05	1.01E-05	2.00E-06	6.32E-07	1.25E-07
5	4.05E-03	7.99E-04	2.53E-04	5.00E-05	1.58E-05	3.12E-06
10	1.62E-02	3.20E-03	1.01E-03	2.00E-04	6.32E-05	1.25E-05
25	1.01E-01	2.00E-02	6.32E-03	1.25E-03	3.95E-04	7.81E-05
50	4.05E-01	7.99E-02	2.53E-02	5.00E-03	1.58E-03	3.12E-04
100	1.62E+00	3.20E-01	1.01E-01	2.00E-02	6.32E-03	1.25E-03
250	1.01E+01	2.00E+00	6.32E-01	1.25E-01	3.95E-02	7.81E-03
500	4.05E+01	7.99E+00	2.53E+00	5.00E-01	1.58E-01	3.12E-02
1,000	1.62E+02	3.20E+01	1.01E+01	2.00E+00	6.32E-01	1.25E-01
2,000	6.48E+02	1.28E+02	4.05E+01	7.99E+00	2.53E+00	5.00E-01
4,000	2.59E+03	5.12E+02	1.62E+02	3.20E+01	1.01E+01	2.00E+00

Reynolds Number Look up Table, dimensionless	Diameter of Pipes in Inches					
Flow, gpm	2	3	4	6	8	12
1	1.40E+03	9.32E+02	6.99E+02	4.66E+02	3.50E+02	2.33E+02
5	6.99E+03	4.66E+03	3.50E+03	2.33E+03	1.75E+03	1.17E+03
10	1.40E+04	9.32E+03	6.99E+03	4.66E+03	3.50E+03	2.33E+03
25	3.50E+04	2.33E+04	1.75E+04	1.17E+04	8.74E+03	5.83E+03
50	6.99E+04	4.66E+04	3.50E+04	2.33E+04	1.75E+04	1.17E+04
100	1.40E+05	9.32E+04	6.99E+04	4.66E+04	3.50E+04	2.33E+04
250	3.50E+05	2.33E+05	1.75E+05	1.17E+05	8.74E+04	5.83E+04
500	6.99E+05	4.66E+05	3.50E+05	2.33E+05	1.75E+05	1.17E+05
1,000	1.40E+06	9.32E+05	6.99E+05	4.66E+05	3.50E+05	2.33E+05
2,000	2.80E+06	1.86E+06	1.40E+06	9.32E+05	6.99E+05	4.66E+05
4,000	5.59E+06	3.73E+06	2.80E+06	1.86E+06	1.40E+06	9.32E+05

The Re is used with a Moody diagram, Fig. 12.2, to determine "f ". For smooth pipes, e.g., plastic, copper, glass, etc., Table 12.1 can be used to obtain the friction factor, f, given the Reynolds number (Re) taken from Table 12.2 or calculated using Eq. 12.8. Table 12.2 can be used to read Re directly for flows between 1 and 4,000 gpm (0.2 to 900 m^3/hr) and pipe diameters from 2 to 12 inches (5 to 30 cm).

Table 12.1 Friction Factor, f, for various Reynolds numbers (Re)

Re	f
2,300	0.042
10,000	0.030
20,000	0.025
50,000	0.021
75,000	0.019
100,000	0.018
200,000	0.016
500,000	0.013
1,000,000	0.012
2,000,000	0.011

note: for Re < 2,300, f = 64/Re

As an easy alternative, many pipe manufacturers and engineering texts (this one included) present frictional head losses for several different types of piping in tabular form, Table 12.3 for PVC Schedule 40 pipe. The input variables are nominal pipe diameter and the flow rate desired. The table than lists the average pipe velocity and the frictional head loss per unit length, usually 100 ft (30 m). The total frictional loss for a system is than calculated by multiplying the table value by the number of unit lengths of pipe run. Again, these tables are based on some assumptions as to pipe characteristics, temperature, and conditions. Since most tables do not consider biofouling, values should be used conservatively.

Example 1

Calculate the friction loss in 200 lineal feet of 4 inch PVC pipe (smooth) flowing at 100 gpm:

Constants: g = 32.2 ft/s^2 and v = 0.0000122 ft^2/s (water at 60°F; see Table 2.1)

1. Cross-section area: $$A=\pi r^2=\pi\times(0.167\ \text{ft})^2=0.0876\ \text{ft}^2$$

2. Velocity:

$$V=\frac{Q}{A}=\left(100\text{gal}/_{\text{min}}\times 1\text{ft}^3/_{7.48\text{gal}}\times 1\text{min}/_{60\text{sec}}\right)\times\frac{1}{0.0876\ \text{ft}^2}=2.54\text{ft/sec}$$

3. Reynolds Number:

$$\text{Re}=\frac{VD}{v}=\frac{2.54\ \text{ft/s}\cdot 0.333\ \text{ft}}{0.0000122\ \text{ft}^2/s}=69{,}300$$

Or using Table 12.2: Re= 70,000

4. From Moody Plot (Fig. 12.2) or Table 12.1

f = 0.019

5. Darcy-Weisbach equation

$$H_L=f\left(\frac{V^2}{2g}\right)\left(\frac{L}{D}\right)$$

$$H_L=0.019\cdot\left(\frac{(2.54\ \text{ft/s})^2}{2(32.2\ \text{ft/s}^2)}\right)\left(\frac{200\ \text{ft}}{0.333\ \text{ft}}\right)$$

$$H_L=1.15\ \text{ft}$$

The same results could be obtained from Table 12.3 for 100 gpm flow rate, 4 inch PVC Schedule 40 pipe. Velocity is 2.56 fps and head loss per 100 ft is 0.58 ft or for 200 ft of pipe, 1.16 ft. In addition to finding the head loss, this table is helpful in choosing pipe size for a given flow rate and assuring that there is sufficient velocity to maintain solids in suspension and avoid scouring of walls and junctions.

12.2 FITTING LOSSES

Energy losses due to fittings are proportional to the dynamic head $(V^2/2g)$, just as the friction losses were. Examining Eq. 12.6 (Darcy-Weisbach equation) suggests that the proportionality constant relating this head loss to the dynamic head, K, is:

$$K = f\frac{L}{D} \tag{12.9}$$

Thus the frictional head loss for any given fitting or transition is found by multiplying the velocity head or dynamic head times the loss coefficient K:

$$H_L = K\left(\frac{V^2}{2g}\right) \tag{12.10}$$

Fittings are assigned a "K" value and the energy loss is simply the product of "K" and the dynamic head. The resistance coefficient K is considered to be independent of the Reynolds (Re) number, as opposed to pipe friction losses, which are dependent upon Re number.

Manufacturers will often specify a flow coefficient for a valve or fitting, C_v, which is related to the friction factor K as follows:

$$C_V = \frac{29.9d^2}{\sqrt{K}} \tag{12.11}$$

$$K = \frac{891d^4}{C_V^2} \tag{12.12}$$

K values for common fittings are listed in Table 12.4. Values for inlets and outlets are shown in Table 12.5. These K values for fittings were produced from manufacturer data given that gives specific frictional loss factors based upon the size of a fitting (f_T) and loss coefficients that are based upon the type of fitting ($FITTING_{constant}$) as follows:

$$K = f_T \times FITTING_{constant} \tag{12.13}$$

Friction loss coefficients can be found in product guides and specification manuals, e.g., "Thermoplastic Valves & Accessories:

Product Guide and Engineering Specifications, 18th Edition", Spears Manufacturing Company, Sylmar, CA; http://www.spearsmfg.com.

Estimating K values accurately depends on the type of fitting, the abruptness of the flow change, the material, and the condition of the internal surfaces. For some fittings, such as valves, there may be considerable variation in losses from one type and manufacture to another. Even for standard fittings, the variation in frictional head losses can be as large as 50%, and thus, pressure loss data from a fitting manufacturer is preferred. The head loss for other types of inline components, such as heat exchangers and UV systems, are usually provided by the manufacturer for the design flow rate.

A general rule of thumb is that you can simply assume every fitting and/or turn in a pipe is equal to one dynamic head, i.e., K = 1.

"Rule of Thumb"
Each fitting will cause one dynamic head of pressure loss, $V^2/2g$.

A second method commonly used for calculating minor head losses uses the concept of Equivalent Length. In this method, the head loss for the fitting is expressed in terms of the length of straight pipe with the same head losses. Tabulated data on fittings is presented in Table 12.6 as a factor (L/D) that is multiplied by the associated pipe diameter to produce and equivalent length of straight pipe (ft or m).

$$Total\ Equivalent\ Length = \left(\frac{L}{D}\right) * \frac{diameter(in)}{12} * number\ of\ fittings$$

or

$$Total\ Equivalent\ Length = \left(\frac{L}{D}\right) * \frac{diameter(cm)}{100} * number\ of\ fittings$$

Thus, the process of estimating total friction head loss in a system consists of first using a diagram of the system to determine the total lengths of each section of pipe with the same diameter and flow rate. Then using the tables provided or the Darcy-Weisbach equation, calculate the head loss per 100 ft and then the head loss in each section of corresponding pipe. Add to this the equivalent length of all valves and fitting in each section, based on the velocity and loss coefficient. Finally, add up the friction losses. For more detailed information on pipe fitting loses and fitting K factors, see Crane Co.'s Technical Paper No. 410.

Example 2

Calculate the dynamic loss due to five 90° elbows of 4 inch PVC pipe (smooth) flowing at 100 gpm:

Constant: $g = 32.2\ \text{ft/s}^2$

1. Pipe Cross-sectional area:

$$A=\pi r^2=\pi\times(0.167\ \text{ft})^2=0.0876\ \text{ft}^2$$

2. Water Velocity:

$$V=\frac{Q}{A}=\frac{\frac{100\text{gal}}{\text{min}}\cdot\frac{1\text{ft}^3}{7.48\text{gal}}\cdot\frac{1\text{min}}{60\text{sec}}}{0.0876\text{ft}^2}$$

V=2.54 ft/s

3. Head Loss coefficient for 90° elbow → K = 0.51 (from Table 12.4) or you could just assume a K =1.

4. Substitution into Eq.12.10:

$$H_L=K\left(\frac{V^2}{2g}\right)$$

$$H_L=0.51\cdot\left(\frac{(2.54\ \text{ft/s})^2}{2(32.2\ \text{ft/s}^2)}\right)$$

$H_L = 0.051$ ft per elbow × 5 elbows

$H_L = 0.26$ ft

Using the concept of equivalent lengths:

$$H_L=\left(\frac{L}{D}\right)*\frac{d}{12}$$

from Table 12.5 , L/D = 30 for 90° elbow:

$$H_L=30*\frac{2}{12}*5\ elbows \qquad H_L=50\ ft\ of\ 4"\ pipe$$

From Table 12.3, the head loss for 4" PVC pipe per 100 ft at 100 gpm is 0.58 ft or H_L is 0.29 ft.

Table 12.3 Frictional Head Losses per 100 ft Schedule 40 PVC Pipe (friction losses for Schedule 80 pipe are provided in the Appendix Table A-20.)

GPM	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)
	1/2 inch			3/4 inch			1 inch			1-1/4 inch			1-1/2 inch			2 inch			3 inch		
1	1.13	2.08	0.90	0.63	0.51	0.22															
2	2.26	4.16	1.80	1.26	1.02	0.44	0.77	0.55	0.24	0.44	0.14	0.06									
5	5.64	23.44	10.15	3.16	5.73	2.48	1.93	1.72	0.75	1.11	0.44	0.19	0.81	0.22	0.09						
7	7.90	43.06	13.64	4.43	10.52	4.56	2.72	3.17	1.37	1.55	0.81	0.35	1.13	0.38	0.17						
10				6.32	20.04	8.68	3.86	6.02	2.61	2.21	1.55	0.67	1.62	0.72	0.31	0.98	0.21	0.09			
15				9.48	42.46	18.39	5.79	12.77	5.53	3.31	3.28	1.42	2.42	1.53	0.66	1.46	0.45	0.19			
20							7.72	21.75	9.42	4.42	5.59	2.42	3.23	2.61	1.13	1.95	0.76	0.33	0.88	0.11	0.05
25										5.52	8.45	3.66	4.04	3.95	1.71	2.44	1.15	0.50	1.10	0.17	0.07
	4 inch																				
30										6.63	11.85	5.13	4.85	5.53	2.39	2.93	1.62	0.70	1.33	0.23	0.10
35										7.73	15.76	6.82	5.66	7.36	3.19	3.41	2.15	0.93	1.55	0.31	0.13
40	1.02	0.11	0.05							8.84	20.18	8.74	6.47	9.43	4.08	3.90	2.75	1.19	1.77	0.40	0.17
				5 inch																	
45	1.15	0.13	0.06										7.27	11.73	5.08	4.39	3.43	1.49	1.99	0.50	0.22
50	1.28	0.16	0.07										8.08	14.25	6.17	4.88	4.16	1.80	2.21	0.60	0.26
60	1.53	0.22	0.10	0.97	0.07	0.03										5.85	5.84	2.53	2.65	0.85	0.37
							6 inch														
70	1.79	0.30	0.13	1.14	0.10	0.04										6.83	7.76	3.36	3.09	1.13	0.49
75	1.92	0.34	0.15	1.22	0.11	0.05										7.32	8.82	3.82	3.31	1.28	0.55
80	2.05	0.38	0.16	1.30	0.13	0.06	0.90	0.05	0.02							7.80	9.94	4.30	3.53	1.44	0.62
90	2.30	0.47	0.20	1.46	0.16	0.07	1.01	0.06	0.03							8.78	12.37	5.36	3.98	1.80	1.78
										8 inch											
100	2.56	0.58	0.25	1.62	0.19	0.08	1.12	0.08	0.04							9.75	15.03	6.51	4.42	2.18	1.94
125	3.20	0.88	0.38	2.03	0.29	0.13	1.41	0.12	0.05										5.52	3.31	1.43
150	3.84	1.22	0.53	2.44	0.40	0.17	1.69	0.16	0.07	0.97	0.04	0.02							6.63	4.63	2.00
													10 inch								
175	4.48	1.63	0.71	2.84	0.54	0.24	1.97	0.22	0.10	1.14	0.06	0.02							7.73	6.16	2.67
200	5.11	2.08	0.90	3.25	0.69	0.30	2.25	0.28	0.12	1.30	0.07	0.03							8.83	7.88	3.41
																12 inch					
250	6.40	3.15	1.36	4.06	1.05	0.45	2.81	0.43	0.19	1.63	0.11	0.05	1.03	0.04	0.02						
300	7.67	4.41	1.91	4.87	1.46	0.63	3.37	0.60	0.26	1.94	0.16	0.07	1.23	0.05	0.02						
350	8.95	5.87	2.55	5.69	1.95	0.85	3.94	0.79	0.34	2.27	0.21	0.09	1.44	0.07	0.03	1.01	0.03	0.01			
400	10.23	7.52	3.26	6.50	2.49	1.08	4.49	1.01	0.44	2.59	0.27	0.12	1.64	0.09	0.04	1.16	0.04	0.02			
450				7.31	3.09	1.34	5.06	1.26	0.55	2.92	0.33	0.14	1.85	0.11	0.05	1.30	0.05	0.02			
500				8.12	3.76	1.63	5.62	1.53	0.66	3.24	0.40	0.17	2.05	0.13	0.06	1.45	0.06	0.03			
750							8.43	3.25	1.41	4.86	0.85	0.37	3.08	0.28	0.12	2.17	0.12	0.05			
1000							11.24	5.54	2.40	6.48	1.45	0.63	4.11	0.48	0.21	2.89	0.20	0.09			
1250										8.11	2.20	0.95	5.14	0.73	0.32	3.62	0.31	0.13			
1500										9.72	3.07	1.33	6.16	1.01	0.44	4.34	0.43	0.19			
2000													8.21	1.72	0.74	5.78	0.73	0.32			
2500													10.27	2.61	1.13	7.23	1.11	0.49			
3000																8.68	1.55	0.67			

(Shaded area recommended flow rates to minimize settlement of solids and avoid scouring of walls and junctions)

Table 12.4 K Values for Variety of Fitting Types

Type of Fitting		Nominal Size of Pipe, inches												
		½	¾	1	1.25	1.5	2	2½ & 3.0	4.0	5.0	6.0	8.0 to 10.0	12 to 16	18 to 24
Standard Fittings														
	Elbows-90	0.81	0.75	0.69	0.66	0.63	0.57	0.54	0.51	0.48	0.45	0.42	0.39	0.36
	Elbows-45	0.432	0.4	0.368	0.352	0.336	0.304	0.288	0.272	0.256	0.24	0.224	0.208	0.192
	T-flow through	0.54	0.5	0.46	0.44	0.42	0.38	0.36	0.34	0.32	0.3	0.28	0.26	0.24
	T-branch off	1.62	1.5	1.38	1.32	1.26	1.14	1.08	1.02	0.96	0.9	0.84	0.78	0.72
Valves														
	Gate	0.22	0.20	0.18	0.18	0.17	0.15	0.14	0.14	0.13	0.12	0.11	0.10	0.10
	Globe & Angle Valve	9.18	8.50	7.82	7.48	7.14	6.46	6.12	5.78	5.44	5.10	4.76	4.42	4.08
	Swing Check	2.03	1.88	1.73	1.65	1.58	1.43	1.35	1.28	1.20	1.13	1.05	0.98	0.90
	Hinged Foot	2.03	1.88	1.73	1.65	1.58	1.43	1.35	1.28	1.20	1.13	1.05	0.98	0.90
	Ball	0.08	0.08	0.07	0.07	0.06	0.06	0.05	0.05	0.05	0.05	0.04	0.04	0.04
	Butterfly (D < 8")	1.22	1.13	1.04	0.99	0.95	0.86	0.81	0.77	0.72	0.68	0.63	0.59	0.54
	Butterfly (D = 10 to 14")	0.95	0.88	0.81	0.77	0.74	0.67	0.63	0.60	0.56	0.53	0.49	0.46	0.42
9 Degree Pipe Bend		0.54	0.50	0.46	0.44	0.42	0.38	0.36	0.34	0.32	0.30	0.28	0.26	0.24
	r/D = 1													
	r/D = 10	0.81	0.75	0.69	0.66	0.63	0.57	0.54	0.51	0.48	0.45	0.42	0.39	0.36
	r/D = 20	1.35	1.25	1.15	1.10	1.05	0.95	0.90	0.85	0.80	0.75	0.70	0.65	0.60
180-Return Bend		1.35	1.25	1.15	1.10	1.05	0.95	0.90	0.85	0.80	0.75	0.70	0.65	0.60
Mitre Bends														
	30 degree	0.22	0.20	0.18	0.18	0.17	0.15	0.14	0.14	0.13	0.12	0.11	0.10	0.10
	45 degree	0.41	0.38	0.35	0.33	0.32	0.29	0.27	0.26	0.24	0.23	0.21	0.20	0.18
	90 degree	1.62	1.50	1.38	1.32	1.26	1.14	1.08	1.02	0.96	0.90	0.84	0.78	0.72
Non-size dependent														
	Pipe Entrance, flush	0.50	0.50	0.50	0.50	0.50	0.50	0.50	0.50	0.50	0.50	0.50	0.50	0.50
	Pipe Entrance, projecting	0.78	0.78	0.78	0.78	0.78	0.78	0.78	0.78	0.78	0.78	0.78	0.78	0.78
	Pipe Exit, all types	1.00	1.00	1.00	1.00	1.00	1.00	1.00	1.00	1.00	1.00	1.00	1.00	1.00

Table 12.5 Minor Loss Coefficients, K, for Inlet, Outlets

Entrance/ Outlet Description		K
Square Entrance		1.0
Rounded Entrance: Radius/Pipe Diameter		
	0.05	0.25
	0.10	0.17
	0.20	0.08
	0.30	0.05
	0.40	0.03
Standard TEE, entrance to minor line		1.8
Screened intake with foot valve		10
without foot valve		5.5
Sudden Expansion , V_1 inlet, V_2 outlet velocity		
$h_m = (1 - V_2/V_1)^2 * V_1^2/2g$		
$h_m = (V_1/V_2 - 1)^2 * V_2^2/2g$		
Sudden Contraction: D inlet, d outlet diameter	$(d/D)^2$	
	0.01	0.5
	0.1	0.5
	0.2	0.42
use V_2 to calculate h_m	0.4	0.33
	0.6	0.25
	0.8	0.15
Check valves: Swing type when fully open		2.5
Ball type		70.0
Lift type		12.0
Exit from pipe into pond		1.0

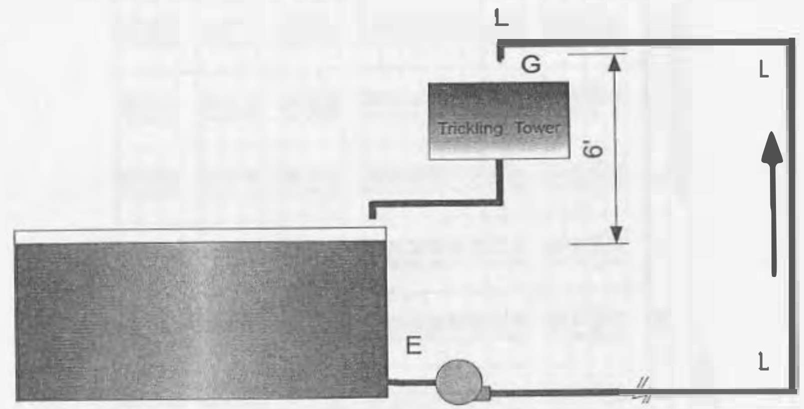
Table 12.6 L/D Ratios for Selected Fittings for Determining Frictional Losses as Equivalent Length of Pipes

Fitting	L/D
Couplings and unions	24
45° elbow	16
90° elbow	30
Closed return bend	50
"Tee" flow along run	20
"Tee" used as elbow (entering branch)	60
Medium sweep elbow	24
Long sweep elbow	20
Globe valve (fully open)	340
Angle valve (fully open)	145
Butterfly valve	20
Check Valve (fully open)	
Swing Type	135
Ball type	150
Lift type	600
Fully open	13
¾ open	35
½ open	160
¼ open	900
Strainer bucket	
With poppet lift-type disc	420
With leather-hinged disc	75

12.3 DESIGN EXAMPLE: HEAD LOSS

Example 3

Assume a required flow of 100 gpm through a side stream CO_2 gas stripping tower. Calculate the total energy loss for a flow from the rearing tank to the water treatment system and gravity flow return to the tank. This system has 100 lineal feet of pipe before the stripping tower, three 90° elbows (L) under pump pressure, an entrance loss into the pump (E) and a gravity return (G); for simplicity assume each elbow has a K = 1.0; system depicted below has a total elevation difference of 6 feet from the top of the water in the tank to the point of discharge above the stripping tower:



For 4 inch PVC pipe:

- **100 lineal feet of pipe @ 100 gpm**

  $H_L = 0.58$ ft (Table 12.3)

- **Three 90° elbows**

$$H_L = K\left(\frac{V^2}{2g}\right)$$

$$H_L = 1.0\left(\frac{(2.55\text{ft/s})^2}{2(32.2\text{ft/s}^2)}\right)$$

$$H_L = .10\ \text{ft}/\text{elbow} \times 3\ \text{elbows}$$

$$H_L = .30\ \text{ft}$$

- **One entrance into the pump**

Entrance $\rightarrow K = 0.5$

$$H_L = K\left(\frac{V^2}{2g}\right)$$

$$H_L = 0.5\cdot\left(\frac{(2.55\ \text{ft/s})^2}{2(32.2\ \text{ft/s}^2)}\right) \quad H_L = 0.05\ \text{ft}$$

- **One gravity return**

Return $\rightarrow K = 1.0$

$$H_L = K\left(\frac{V^2}{2g}\right)$$

$$H_L = 1.0\cdot\left(\frac{(2.54\ \text{ft/s})^2}{2(32.2\ \text{ft/s}^2)}\right)$$

$$H_L = 0.10\ \text{ft}$$

Total energy loss $= \sum H_L + \Delta Z$

$= \sum(0.58 + 0.15 + 0.05 + 0.10) + 6\ ft$

$= 6.88\ ft\ (3.03\ psi)$

Notice that in this particular example that most of the head loss is in the elevation difference, followed by the length of pipe. There are small losses due to the entrance and return. Overall, this was a good choice of pipe size with a flow velocity adequate to maintain any solids in suspension and avoid any scouring of pipe walls. It is important to note that neither the striping tower nor the piping downstream from it have any effect on the required pumping head. This is because the flow was open to the atmosphere at the point the water was released to the top of the stripping unit.

<u>For 3 inch PVC pipe:</u>

Here we present the previous example with a 3 inch pipe to demonstrate the effect of a reduced pipe diameter.

- **100 lineal feet of 3 inch pipe @ 100 gpm**

  $H_L = 2.18$ ft(Table 12.3)

- **Three 90° elbows** $\rightarrow K = 1.0$ (is often used for simplicity, depending on the degree of accuracy required)

$$H_L = K\left(\frac{V^2}{2g}\right)$$

$$H_L = 1.0\cdot\left(\frac{(4.42\ \text{ft/s})^2}{2(32.2\ \text{ft/s}^2)}\right) = 0.30\ \text{ft}$$

$$H_L = 0.30\ \frac{\text{ft}}{\text{elbow}}\cdot 3\ \text{elbows}$$

$$H_L = 0.90\ ft$$

- **One entrance into the pump**

Entrance $\rightarrow K = 0.5$

$$H_L = K\left(\frac{V^2}{2g}\right)$$

$$H_L = 0.5\cdot\left(\frac{(4.42\ \text{ft/s})^2}{2(32.2\ \text{ft/s}^2)}\right)$$

$$H_L = 0.15\ \text{ft}$$

- **One gravity return**

Return $\rightarrow K = 1.0$

$$H_L = K\left(\frac{V^2}{2g}\right)$$

$$H_L = 1.0 \cdot \left(\frac{(4.42 \text{ ft/s})^2}{2(32.2 \text{ ft/s}^2)}\right)$$

$$H_L = 0.30 \text{ ft}$$

Total energy loss $= \sum H_L + \Delta Z$

$= \sum (2.18 + .9 + 0.15 + 0.30) + 6\,ft$

$= 9.53\,ft\ (4.19\,psi)$

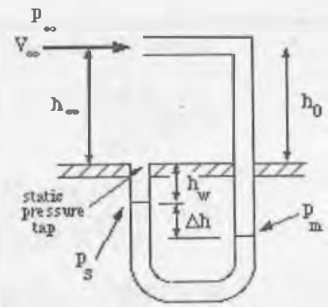
Notice how the total head loss is now 9.53 ft for the 3 inch pipe, compared to only 6.88 ft for the 4 inch pipe. In designing the circulation system, cost saving upfront for PVC pipe of smaller then required diameter might save some capital costs, but in the long run, the operational costs to pump against the increased head, as well as the larger pump size will most likely offset this savings.

For systems with the same velocity throughout, i.e. not change in piping diameter, one can just sum the "K" values and then multiply times the dynamic head; note that the friction loss can be represented as a "K" value by multiplying the friction factor, f, times the relative length (L/D) (Eq. 12.9).

12.4 Measurement of Flow

Fluid flow can be measured either directly to determine actual flow for a given time interval, such as with a bucket and stopwatch, or indirectly by measuring some variable that is directly related to the flow rate, such as pressure differential across an orifice plate. There are numerous devices available for measuring flow rate in pipelines, such as orifice plate meters, propeller meters, electromagnetic flow meters, ultrasonic flow meters, and rotameters. One can obtain a device that is appropriate for specific circumstances and which provides the degree of accuracy required at a reasonable cost. Flow rates can also be estimated from open pipe discharges by relating the height of the water jet from a vertical pipe or the trajectory of a horizontal pipe.

Pitot-Static Gauge or Tube



You can determine the velocity in a flowing pipe by inserting a Pitot-Static Gauge. This device consists of a wall tap into the pipe that measures the static pressure energy (elevation + pressure energy) and a small tube placed to face into the flow that measures total pressure (elevation + pressure + kinetic energy). Generally the wall or static energy pressure tap is built into the pitot tube itself. The difference in these two energy measurements is the kinetic energy only (Δh). This can be seen be rearranging and solving for velocity from Eq. 12.4:

$$V = C \times 2.315\sqrt{\Delta h_{inches}} \qquad (12.14a)$$

$$V = C \times 0.443\sqrt{\Delta h_{cm}} \qquad (12.14b)$$

where V is in ft/s (m/s)
Δh is in inches water gauge (cm water)
C = 0.95 to 1.00 for pitot tube
C = 0.60 for sharp edged orifice

Example 4

What is the velocity in a pipe where the pitot tube indicates a pressure difference ($p_m - p_s$) of 12 inches (30.5 cm) water gauge.

IP units

$$V = 1.0 \cdot 2.315 \cdot \sqrt{12} = 8.02\ ft/s$$

SI units

$$V = 1.0 \cdot 0.443 \cdot \sqrt{30.5} = 2.45\ m/s$$

Venturi Tube

Venturi tube meters estimate flow based upon the change in pressure along a pipe that is specifically reduced in cross sectional diameter so that the fluid velocity is increased which causes a corresponding decrease

in static pressure (see Eq. 12.4 and Fig. 12.3). There are commercially available venturi meters that can be installed in a straight section of pipe to obtain the pressure measurements. Knowing the pressures at the upstream (P_1) and in the throat section (P_2), the velocity can be calculated:

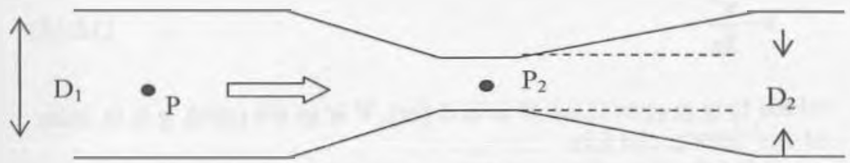


Figure 12.3 Venturi meter showing pertinent variables.

$$V_2 = \frac{C\beta\sqrt{(P_1 - P_2)}}{\sqrt{1-\left(\frac{D_2}{D_1}\right)^4}} \qquad (12.15)$$

where C = 1.00 for perfect venturi (manufacturer specified, generally around 0.98 to 0.99) and dimensions are in cm or inches
P_i = pressure terms in kPa, psi or inches water gauge (velocity is predicted in cm/s or ft/s)
β = 2.312 for US units with pressure in inches water gauge
12.18 for US units with pressure in psi gauge
1.414 for SI units with pressure in kPa[b]

Example 5

Calculate the velocity in a venturi meter (assume venturi coefficient of 1.00) if the venturi has a throat diameter of d = 2 inches (5.08 cm) and the main pipe has a diameter of D = 4 inches (10.2 cm) if the pressure differential between the main pipe and the venturi is 12 inch water gauge (2.983 kPa).

IP Units

[b] 1 inch water gauge pressure is 0.2486 kPa

$$V_2 = \frac{1.00 \cdot 2.312\sqrt{12}}{\sqrt{1-\left(\frac{2}{4}\right)^4}} = 8.27\ ft/s$$

SI Units

$$V_2 = \frac{1.00 \cdot 1.414\sqrt{2.983}}{\sqrt{1-\left(\frac{5.08}{10.2}\right)^4}} = 2.52\ m/s$$

ORIFICE PLATE METER

Equation 12.15 can also be used to predict the velocity in a pipe where an orifice plate with a smaller hole of diameter "D_2" has been inserted. The C coefficient is approximately 0.60 for sharp-edged orifices when the ratio D_2/D_1 is less than 0.3 (see ASHRAE Fundamentals for more specific information where C is related to Reynolds number and D_2/D_1).

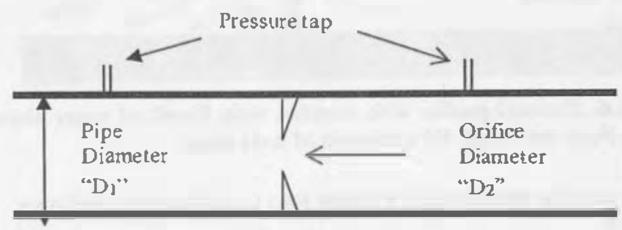
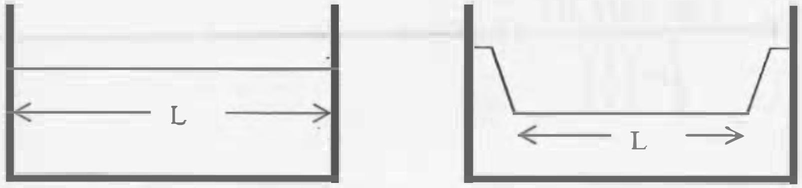


Figure 12.4 Depiction of a pipe with a sharp edged orifice plate.

WEIRS

Weirs are essentially "dams" that are placed in a channel that obstruct the flow, thereby creating a crested flow over the weir. The height of this crest of fluid can be measured and then correlated with a channel flow rate. Use of open channels in recirculating aquaculture systems (RAS) are handy as they are easy to clean and do not plug. The two most common weirs are either rectangular flat weirs or Cippoletti weirs (see Fig. 12.5). Other weir calculations are presented by Lawson (1995) and Piper et al. (1982).



(a) (b)

Figure 12.5 Weir Types: a) Rectangular and b) Cipolletti.

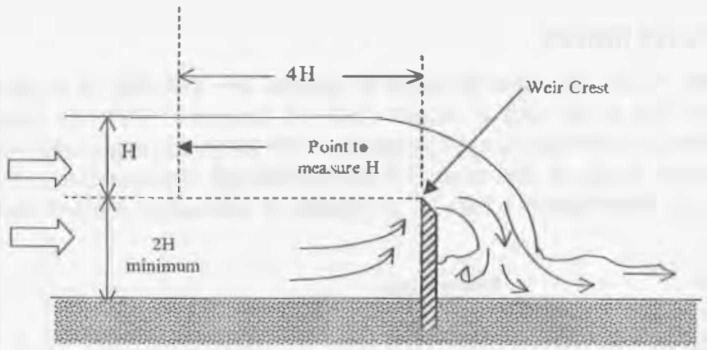


Figure 12.6 Channel profile with inserted weir. Depth of water above weir crest, H, is measured 4H upstream of weir edge.

Channel flow can be predicted for these two weir types as follows:

Rectangular or Cippoletti

$$Q(\text{gpm or L/s})=\beta\, LH^{1.5} \qquad (12.16^{c})$$

[c] Equations 12.16 and 12.17 are for a properly operating rectangular weir with suppressed end contractions, i.e. a weir with a crest equal to the channel width. End contractions are corrected by assuming effective width of crest length is reduced by 0.1H at each end or $L_{effective} = L - 0.2H$.

In the case that there is significant kinetic energy, h, in the approach velocity at the weir, then use the following equation:

$$Q=\beta\, L((H+h)^{1.5}-h^{1.5}) \qquad (12.17^{d})$$

$$h=\frac{V^2}{2g} \qquad (12.18)$$

where Q is in gpm (L/s), H is in ft (m), V is in ft/s (m/s), g is in units of ft/s^2 (m/s^2), and β is:

- 1,511 for Cippoletti weir US units (1,857 SI Units)
- 1,495 for rectangular weir US units (1,837 SI Units)

Example 6

Calculate the flow rate from a Cippoletti weir if the height above the weir crest (4H upstream) is 10 inches (0.254 m) and the weir length is 1.0 ft (0.305 m) and the approach velocity is negligible.

IP[e] Units

$$Q(gpm) = 1{,}512 \cdot 1.00\,ft \cdot (10/12\,ft)^{1.5} = 1{,}150\ gpm$$

SI Units

$$Q(L/s) = 1{,}857 \cdot 0.305m \cdot (0.254m)^{1.5} = 72.54\ L/s$$

Vertical Pipe

Predicting the flow from an open ended round pipe pointed "straight up", Fig. 3.7, can be done using Eqs. 12.19 and 12.20 (more in depth treatment of this method is provided by Lawson, 1995):

[d] Equation 12.17 shows a correction term for approach kinetic energy, h. This was first introduced in 1852 and became known as the Francis formula. It is based upon data that was for channel widths from 8–10 ft (2.4 to 3.0 m) and for hydraulic heads (H) of 0.6–1.6 ft (0.2–0.8 m) and kinetic energies of 0.2–1.0 ft (0.06–0.3 m). Published in the Handbook of Hydraulics by E.F. Brater and H.W. King, 1918.

[e] IP is English or Inch-Pound system.

IP units

$$Q(gpm)=5.39\times D^2H^{0.47} \tag{12.19}$$

SI units

$$Q(L/s)=0.034\times D^2H^{0.47} \tag{12.20}$$

Graphic representations of Eq. 12.20 is represented in Figs. 12.8 and 12.9.

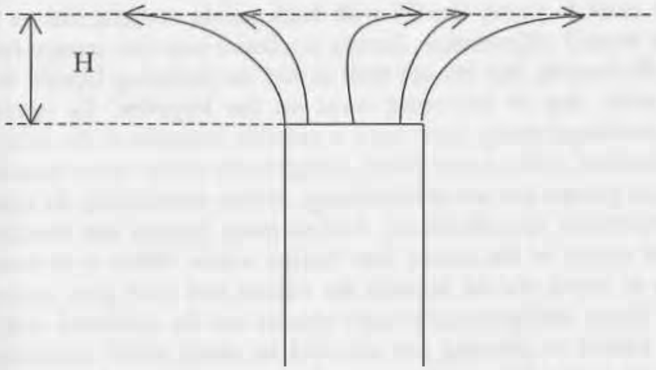


Figure 12.7 Flow from an open ended round pipe pointed "straight up" illustrating the head created "H".

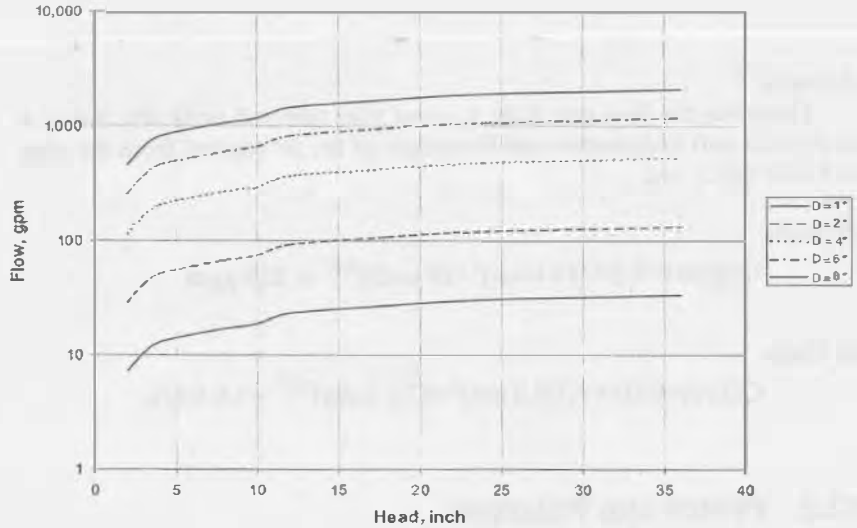


Figure 12.8 Flow estimate for a vertically oriented pipe (from Eq. 12.19).

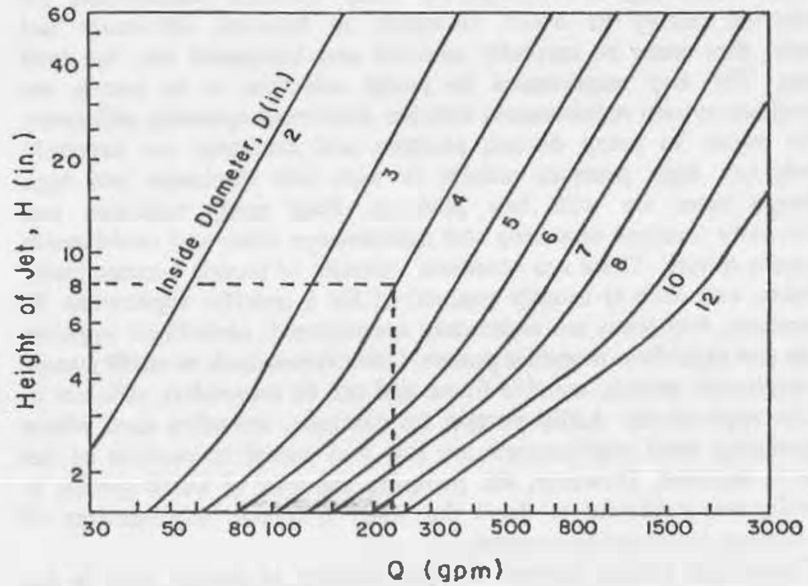


Figure 12.9 Flow estimate for a vertically oriented pipe (from Eq. 12.19) (from US Department of the Interior, 1967).

Example 7

Calculate the flow rate from a round pipe oriented vertically that is 4 inch (10.2 cm) in diameter and the height of the jet emitted from the pipe is 8 inch (20.3 cm).

IP Units

$$Q(\text{gpm})=5.39\times(4\ \text{inch})^2\times(8\ \text{inch})^{0.47}=229\ \text{gpm}$$

SI Units

$$Q(\text{L/s})=0.034\times(10.2\ \text{cm})^2\times(20.3\ \text{cm})^{0.47}=14.5\ \text{L/s}$$

12.5 PUMPS AND PUMPING

All RAS employ some kind of pump to move water to a higher elevation or to increase the overall system pressure for filtration, aeration, degassing, etc. Pumps are fairly efficient mechanisms for transferring energy to water. However, to function efficiently and reliably, they must be carefully selected and integrated into the total system. The key requirement in pump selection is to match the aquaculture system requirements with the maximum operating efficiency of the pump. In pump design, pressure and discharge are inversely related, i.e., high pressure usually is with low discharge and high discharge rates are with low pressure. Poor pump selection can significantly increase operating and maintenance costs and could result in system failure. There are countless varieties of pumps commercially available, and each is usually engineered for a specific application. In aquaculture, two types are commonly encountered: centrifugal impeller pumps and axial flow propeller pumps. Other types, such as airlift pumps and diaphragm pumps, are also found and can be reasonably efficient in specific applications. Airlift pumps, for example, are often used where the pumping head requirements are low and partial re-aeration of the water is required. However, the pumping capacity of airlift pumps is generally not sufficient to drive the water treatment requirements of high-density recirculation systems.

Centrifugal pumps account for the majority of pumps used in the aquaculture industry. They are sometimes called radial flow pumps, because water enters the pump at the eye, or center of the impeller and is driven outwards by centrifugal force at right angles to the axis of rotation. The centrifugal pump characteristics are determined to a large degree by the impeller design. The impeller generally has the form of a disk with a series of curved, raised vanes radiating from the center. The spacing between the impeller and the volute together with the impeller geometry, limits the size of solids that can pass through the pump. Totally enclosed impellers have faceplates that completely enclose the waterways between the vanes on both sides. In open and semi-enclosed impeller pumps, the clearances between and around the vanes are large, allowing large solids to pass through the pump. Open and semi-enclosed impellers are used to pump liquids with high solids content, but at the cost of lower overall efficiencies. Totally enclosed impeller pumps have the highest efficiencies, but are not well suited for pumping liquids with suspended solids, due to increased wear on the impeller. To operate properly, a centrifugal pump must have a positive pressure at the inlet or be directly plumbed with a water filled, airtight pipe to the water source.

Centrifugal pumps are not self-priming, unless specifically so stated in the manufacturers specifications. Self-priming pumps are designed with a special cavity in the volute that retains water. When it is turned on, it is able to expel the air in both the volute and inlet pipe and re-prime itself. These self-priming pumps should not be confused with a simple filter basket or priming pot attached to many small centrifugal pumps. A pump with a filter basket and a foot valve will retain its prime, as long as no air enters the system. However, if any air enters the systems, due to a leaky foot valve for example, the impeller will spin in an empty volute once the water in the filter basket has been emptied out.

The best way to prevent loss of prime is to locate the pump below the surface of the water level being pumped. This is commonly referred to as a flooded suction, since when the pump is off, it's still flooded with water. If the pump must be located higher than the surface of the water, then a foot valve should be installed at the bottom of the suction pipe, alone with an inlet strainer. Some form of priming access, either a priming pot, filter basket or TEE fitting should be placed at the inlet to the pump, to allow the suction pipe to be completely flooded with water. Once flooded and sealed at the top and bottom, the pump should not lose prime when turned off. It is important not to restrict the flow of water into a centrifugal pump with a valve or extensive plumbing. This can result in a condition called cavitation, where some or all of the fluid has turned into water vapor. Cavitation can damage the shaft or impeller and significantly reduces or eliminates all flow from the pump. Burnt out pump motors can result.

Axial flow pumps are designed to pump water efficiently at low head and high discharge. The pumping element of an axial flow propeller

pump consists of a revolving propeller, similar in appearance and function to a boat propeller. The propeller is located inside of a pipe or shaft that serves as a conduit to convey the water. The propeller is driven by either a submersible motor or a remote-mounted motor linked by a drive shaft. The water flows in essentially a straight line along the pump axis keeping friction and turbulence to a minimum. The propeller of an axial flow pump must be submerged in the water. Axial flow pumps can be extremely efficient at moving large volumes of water to modest head levels, e.g., 4.6 to 9.2 m (15 to 30 feet). In addition, they are able to tolerate some small debris and small solids.

Selecting a pump requires knowledge of at least the following factors:

- Total head required
- Discharge flow required
- Suction lift required
- Liquid to be pumped and its characteristics (freshwater, saltwater, solids)
- Continuous or intermittent pumping
- Power source available (single or three phase power, diesel or gas engine)
- Space, weight, and similar limitations
- Special requirements

The term "head" is commonly used to express the pressure at some point in terms of elevation (feet or meters). The following expressions of "head" are often found in the industry to describe performance and design parameters for systems:

- **Static Head:** the hydraulic pressure at a point in the water when the water is at rest

- **Friction Head**: the loss in pressure or energy due to frictional losses in flow

- **Velocity Head:** the energy in a fluid due to its velocity

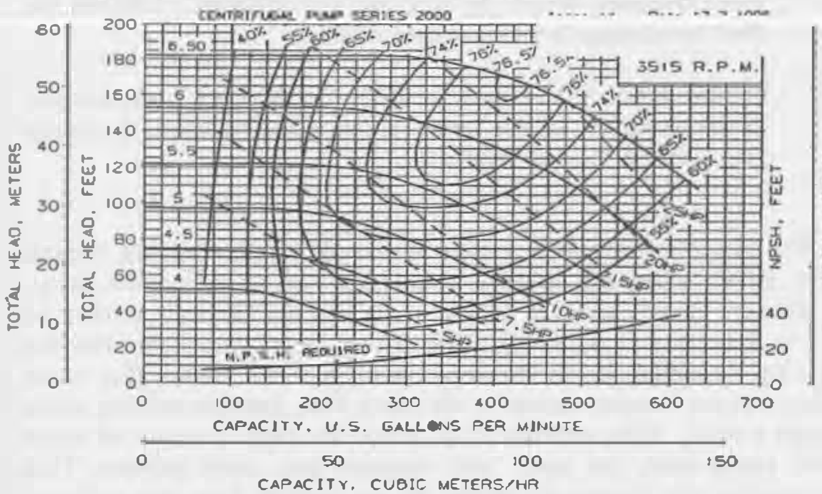
- **Pressure Head:** a pressure measured in equivalent head units

- **Discharge Head:** the output pressure of a pump in operation

- **Total Dynamic Head:** the total pressure difference between the inlet and outlet of a pump in operation

- **Suction Head:** the vertical distance from the static water surface to the centerline of the pump intake, positive when the pump inlet is below the water surface

The total suction head or suction lift should also include the friction losses, minor losses, and velocity head on the suction side of the pump, but these are usually small in relation to the suction lift and can often be ignored. The water in the suction line of a pump is being pulled into the pump by the difference in the atmospheric pressure on the free water surface and the vacuum created in the pump inlet, just like sucking water through a straw. If the pressure drops below the vapor pressure of water in the pump inlet, the water will vaporize into small bubbles. This phenomenon also creates cavitation. As the bubbles form and collapse, they can stress the metal pump housings and impellers, and pitting can occur on their surfaces. Cavitation is easy to recognize, since it sounds like the pump is full of gravel or has a rattling sound. Cavitation reduces the pumps overall efficiency and can severally damage a pump and impeller if not corrected.



Impeller Code	A	B	C	D	E	F
Size	6.5	6	5.5	5	4.5	4
Standard HP	25	20	15	10	7 ½	5

Figure 12.10 Performance curve of a centrifugal pump (Courtesy of Goulds Pumps).

Theoretically, the greatest such lift possible is 10.36 m (34 feet) at sea level, but practical limits reduce this to between 4.5 and 6.1 m (15 and 20 feet) for reasonably efficient operations. The possible lift for different elevations is shown in Table 12.7. The net positive suction head required ($NPSH_{required}$) to prevent cavitation is a function of the pump design and speed, and is usually specified by the manufacturer. A typical performance curve is shown in Fig. 12.10. This figure shows the total head that the pumps operate against (ordinate axis), the flow rate of the pump is on the abscissa axis; the $NPSH_{required}$ is shown on the right hand side of the performance curves on an ordinate axis. Efficiencies are shown on the graph for the different operating points of the pump.

The $NPSH_{required}$ must be less than the $NPSH_{available}$ or:

$$NPSH_{available} \geq NPSH_{required} \qquad (12.21)$$

Where $NPSH_{available}$ is defined as the atmospheric pressure less friction losses on the suction side of the pump, the suction lift, and the vapor pressure of the water. The atmospheric pressure is given in Table 12.7 and the vapor pressure is given in Table 2.1.

Table 12.7 Standard Atmospheric Pressure at Different Altitudes

Altitude		Atmospheric Pressure				
m	ft	mm Hg	kPa abs	psia	m H_2O	ft H_2O
-200	-656	778	103.7	15.0	10.6	34.8
0	0	760	101.3	14.7	10.3	33.8
200	656	742	98.9	14.3	10.1	33.1
400	1,312	725	96.6	14.0	9.9	32.5
600	1,968	707	94.3	13.7	9.6	31.5
800	2,625	690	92.0	13.3	9.4	30.8
1,000	3,281	674	89.8	13.0	9.2	30.2
4,000	13,123	462	61.6	8.9	6.3	20.7

The efficiency of a pump is defined as the ratio of work output or water horsepower (WHP) and the energy input used to operate the pump (Brake Horse Power, BHP):

$$WHP = \frac{Q \times TDH \times \Omega_{sg}}{\beta} \qquad (12.22)$$

$$E_{pump}(\%) = 100\frac{WHP}{BHP} = \frac{output}{input} \qquad (12.23)$$

β = 6,116 for WHP in kW and Q in L/min and TDH in m
0.102 for WHP in kW and Q in m^3/s and TDH in m
3,960 for WHP in HP and Q in gpm and TDH in ft
8.81 for WHP in HP and Q in cfs (ft^3/s) and TDH in ft

The Total Dynamic Head (TDH) is the sum of the static lifts from the water source to the discharge point, and all the frictional losses from the inlet suction side to the discharge side. The velocity head is often negligible but should be considered when extremely accurate values are needed. The frictional losses include piping, fittings, and processing equipment (for example UV, heating, and oxygenation systems) on both the suction and the discharge sides of the pump. Since the frictional losses are a function of the flow rate, the TDH must be calculated over the range of anticipated flows. Considerations should be made of

anticipated changes in operations and management, biofouling, or system configurations.

Once the system is initially designed and the TDH is estimated at the desired flow rate, then a pump must be selected that minimizes pumping costs by operating at or near its maximum efficiency to deliver the desired flow. This is accomplished by examining the performance data for the various pumps under consideration. Performance data is usually presented in graphical form, although for small pumps, tables of head versus discharge are usually provided. Each pump has its own unique set of characteristic curves, determined by the engineering design of the impellers, casing and to some extent by its age and wear of internal parts. A typical performance curve for a centrifugal pump was previously shown in Fig. 12.10 and performance data is given for a small submersible centrifugal pump in Table 12.8.

The flow of water is directly related to the TDH with maximum water flow occurring at the lower TDH values. Some pumps will have relatively flat responses in pumping rate to changes in TDH while other pumps are extremely dependent upon the TDH that the pump is operating against.

Each aquaculture system will have its own system operating curve. The system operating curve is constructed by computing the total dynamic heads required in the proposed system to deliver a range of specific flow rates. Once a system operating curve is constructed, then the manufacturer's pump characteristic curves can be imposed on the system curve and where the two curves intersect defines the system's operating point. Ideally, you try to chose a pump such that the intersection of the operating and system curves are as close as possible to the maximum operating efficiency of the pump (see Fig. 12.10). Note in this figure, that for a 15 HP pump, the maximum efficiency will occur at an operating point of approximately 310 gpm (70 m^3/hr) at a TDH of 110 ft (33.6 m). However, if you operated this same pump at a TDH of 60 ft (18.3 m), you would obtain a flow rate of 520 gpm (118 m^3/hr). Now, do not be totally misled by thinking that the only objective is to maximize pumping efficiency. In the previous example, it may be advantageous for you to operate the pump at a lower TDH and obtain the higher flow rate, particularly if your system design indicated that you only required approximately 15 to 30 feet (50 to 60 feet) of TDH. The principles of system operating curve and pump operating characteristics are demonstrated in Fig. 12.11.

Table 12.8 Performance Data for a Submersible Sump Pump

	GPM @ Total Feet of Head					
HP	5 ft	10 ft	15 ft	20 ft	25 ft	30 ft
½	58	37	18	0	0	0
¾	65	47	31	15	0	0
1		58	43	29	15	0
1.50		81	63	48	32	16
1.75		94	78	62	47	32

Data for an ABS Stainless Steel Submersible pump

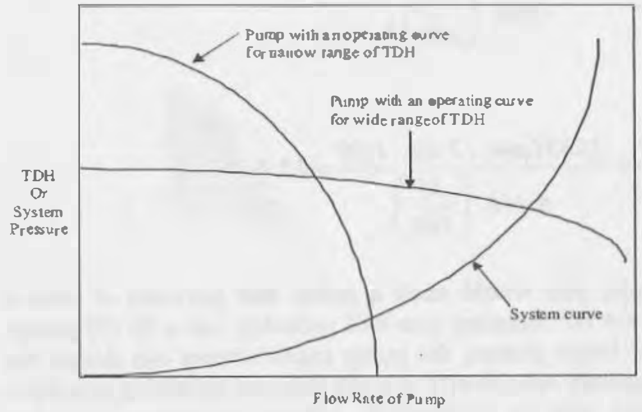


Figure 12.11 Pump characteristic curve and a system operating curve.

If a range of discharges were required, the pump selected would be capable of providing the maximum discharge. The pump would have the flattest possible efficiency curve over the necessary operating ranges, with the maximum efficiency at the operating point used the greatest amount of the time. Moreover, for large, commercial operations, this would be the design engineer's method of selection, plus some practical experience with other systems and pumps

In the real world, selecting a pump for small systems is usually done by estimating the overall system's total dynamic head and flow requirements. Then, the characteristic curves for several readily available pumps that can provide the flow rate at the required head are reviewed. In some cases, efficiency data for pumps is also provided, but normally not. So a guess is made as to the maximum efficiency, usually just to the right of the midpoint on the characteristic curve. A pump is chosen after

considering other factors such as capital cost, operating cost, size, color, discounts, and availability.

Example 8

Calculate the brake horsepower required to operate a pump that had a pumping efficiency of 55%, operates against a TDH of 40 ft and pumps at a flowrate of 1,000 gpm.

IP

$$BHP = \frac{WHP}{E_{pump}} = \frac{1{,}000\,gpm \cdot 40\,ft \cdot 1.00}{3{,}960 \cdot \left(\frac{55}{100}\right)} = 18.4\,HP$$

SI

$$BHP = \frac{WHP}{E_{pump}} = \frac{3{,}785\,Lpm \cdot 12.2\,m \cdot 1.00}{6.116 \cdot \left(\frac{55}{100}\right)} = 13.7\,kW$$

In this example, you would need a pump that provides at least a power rating of 18.4 HP meaning you will probably use a 20 HP pump. In some cases, for larger pumps, the pump manufacturer can design the pump for you to operate specifically at a pre-selected operating condition and then the pump will be designed to achieve maximum operating efficiency for these specific conditions. This is particularly relevant to aquaculture applications where generally we need high flow rates against fairly low operating heads. In general, such pumps for large applications have not been available. The closest companion industries are irrigation pumps.

Sump Sizing

Don't forget to size a receiving sump adequately so that a flooded suction pump remains submerged or the pump remains submerged where necessary. A rule of thumb is 1055 m^3/d per m^2 or 18 gpm per ft^2 coupled with a hydraulic retention time of 3 to 5 minutes. Alternatively, take your total flow rate times the 3 to 5 minutes retention time to obtain volume and then selecting a sump water depth will define the cross sectional area of the sump. What you are trying to avoid is the sump being pumped "dry" during startup when an inactive pump reactivates. In other words, the sump under normal operation will have a lower water level than when the system shuts down and receives the water draining from the

various devices and tanks that have water at higher elevations. Don't forget that the sump pump side walls should (whenever possible) be as high as the normal level of the water in the fish tank. This will prevent an inadvertent draining of your fish tank when something goes awry in your sump pump operation.

12.6 Airlift Pumps[f]

In principle and theory, airlift pumps will by far move the most water per unit of energy supplied. However, airlift pumps have the following disadvantages:

- Limited application to create water lift or elevation change, e.g., 10 to 15 cm.
- Reductions in flow rates due to fouling of the air distribution mechanism, particularly for conventional sintered glass air stones
- Complete loss of water pumping when water elevation changes cause excessive requirements for water lift, e.g., water level dropped in tank where the unit is placed
- Reductions in water flow are not "obvious" to casual observer
- Energy efficiency is highly related to very closely matched requirements between centrifugal blower and the airlift requirements

All of the five disadvantages listed above can be eliminated with effective management, maintenance, and initial design of the overall system. Practically speaking, airlifts are problematic. In particular, the authors became discouraged with their application because airlifts are constrained to low water head differentials. We would suggest that airlifts are most appropriate in low density fish applications. Figure 12.12 demonstrates the important variables to consider when designing an airlift pump. In the appendix, the software program AIRPUMP is included that predicts the operating performance of such pumps.

The airlift pump has been in practical use as a pumping device for many decades. A gas, usually air, is injected at the base of a submerged riser tube. As a result of the gas bubbles suspended in the fluid, the average density of the two-phase mixture in the tube is less than that of the surrounding fluid. The resulting buoyant force causes a pumping action.

[f] For downloadable software to predict airlift pump performance, see: www.bee.cornell.edu/aqua

Complicating the prediction of the hydrodynamics of the airlift is the fact that there are three flow patterns possible, Fig. 12.13. When the initial bubble size is much smaller than the tube diameter and the gas void ratio is below about 25%, the bubble flow pattern results. Small bubbles are distributed over the pipe cross section. Bubbles remain close to their initial size, and there is little interaction between single bubbles. When gas void fraction exceeds 25%, coalescence occurs and large gas bubbles or "slugs' form.

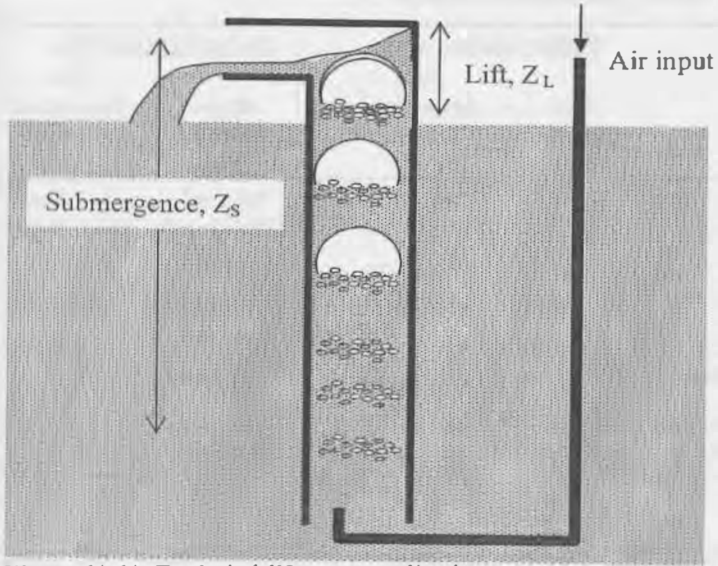


Figure 12.12 Typical airlift pump application.

In slug flow, the gas phase is contained in large bubbles that nearly span the tube and range in length from the tube diameter to several times this value. These are referred to as gas slugs or Taylor bubbles. The liquid filling the space between the Taylor bubbles is referred to as the liquid slug. The liquid between the Taylor bubbles and the tube wall is referred to as the liquid film. The slug flow regime has been found to occur only when the riser tube diameter is below 25 mm (1 inch).

The bubbly-slug flow regime is most commonly encountered in airlift operation. In the bubbly-slug flow regime, small bubbles are found suspended in the liquid slug between the Taylor bubbles. The presence of these bubbles is due to the region of extreme turbulence encountered at the tail of the Taylor bubble. Small bubbles are broken off of the tail of

the Taylor bubble and dispersed in the liquid slug. The bubbly-slug regime occurs in tubes with diameter greater than 25 mm (1 inch) when the gas void ratio is above about 25% regardless of the initial bubble size.

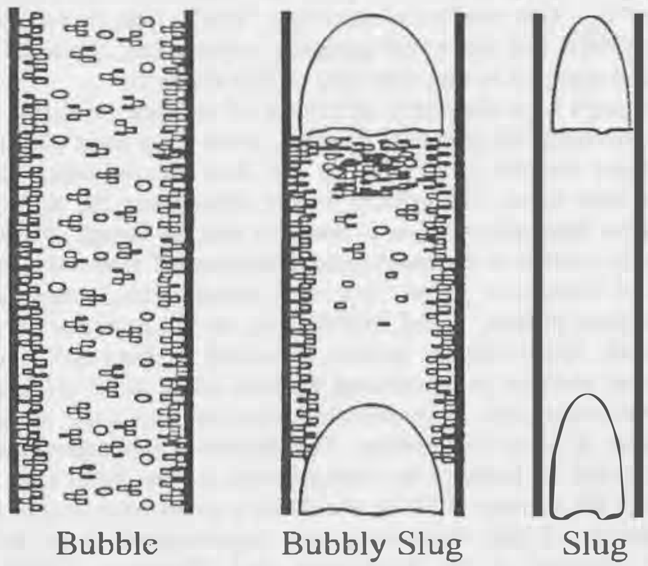


Figure 12.13 Three flow patterns encountered in airlift pump operation.

The gas concentration in the tube is a function of the submergence ratio and the amount of air being introduced into the pump. The submergence ratio is defined as the fraction of the tube that is submerged below the level of the water being pumped (see Fig. 12.12). Airlift pumps must be submerged at least 50% for any pumping action to take place. The pumping efficiency is defined as the useful work done raising the water divided by the work required to compress the air. Maximum pumping efficiencies are obtained for submergence ratios of approximately 80% depending on tube size and flow pattern. Maximum pumping rates are obtained with 100% submergence. Although there is no useful pumping work done when the pump is 100% submerged (since there is no lift), this configuration can be used to mix and aerate tanks and ponds.

The length to diameter ratio (Z:D) of the riser tube can have a significant effect on airlift pump performance (note that Fig. 12.12

depicts Z_s and Z_L where $Z = Z_s + Z_L$). The length to diameter ratio is defined as the length of the riser tube divided by its diameter, with both measurements in the same units, e.g., a 10 meter riser tube with a diameter of 10 centimeters has a length to diameter ratio of 100. When Z:D is below 50 the two phase flow patterns will not have sufficient time to develop fully. This results in increasing "slip" of the water past the rising gas bubbles and decreased pumping volume and efficiency. It is therefore advantageous to keep the ratio of Z:D above 50.

Airlift pumps have the added advantage of aerating the water in the process of providing the pumping function, since air is used to drive the pump. The gas transfer is affected by the flow rate of water and air through the riser tubes. The critical factors influencing the rate of gas transfer are the Reynolds number of the flow and the length of the tube. The Reynolds number is a dimensionless measure of flow velocity and the degree of turbulence in the flow. Gas transfer is not significantly affected by flow pattern, initial bubble size, or the presence of dilute organic wastes. Some caution must be exercised in using airlift pumps, since the water and thus gases entering the base of the airlift column may be above saturation due to hydrostatic pressure; this may cause gas bubble disease in some fish species. The degree of gas supersaturation can be controlled by limiting the submergence of the airlift riser. This will also limit the amount of lift available for a given tube length. An in depth treatment of the theoretical and experimental basis for the AIRPUMP program is in Reinemann and Timmons (1987) and Reinemann et al. (1990).

12.7 Design Example - Circulation

Table 5.14 is reprinted below to summarize the engineering design at this stage, i.e. pump selection. The final pump specifications will depend upon several other design selections such as the type of solids capture device, biofilter, or oxygen transfer system. For example, a microscreen filter is usually gravity fed from the combined flows from the center drains or the discharge (overflow) of a radial flow clarifier/separator, while a propeller-washed bead filters (PBF) requires a collection/pump sump and a pump. As previously mentioned, one advantage of the Moving Bed Biofilter is the low head loss across it. One disadvantage is the difficulty in finding an energy efficient pump that will provide the necessary flow rate at this low head requirement. Finally, the transfer efficiency and discharge oxygen concentration for a Speece cone is dependent both on

the flow rate and the operating pressure, requiring careful selection of the pump. In addition, since most of the pumps will be operated continuously, they need to be as energy efficient as possible to reduce the overall operating cost. This is an example of where a higher upfront capital cost can save money by lowering the long term variable costs (pumping energy).

Table 5.14. Summary of Design Total Volume and Design Flows for the Two Design Scenarios

	Design Scenario One		Design Scenario Two	
	Two Juvenile Pod	Five Fingerling/ Growout Pods	Single Juvenile Pod	Two Fingerling/ Growout Pods
Pod Total Volume:	32.0 m^3 (8,465 gal)	57.2 m^3 (15,110 gal)	96.1 m^3 (25,400 gal)	143.8 m^3 (38,000 gal)
Total Flow:	90.8 m^3/hr (400 gpm)	90.8 m^3/hr (400 gpm)	273 m^3/hr (1200 gpm)	227 m^3/hr (1000 gpm)
Center Discharge:	22.7 m^3/hr (100 gpm)	22.7 m^3/hr (100 gpm)	68.3 m^3/hr (300 gpm)	45.4 m^3/hr (200 gpm)
Side-wall Discharge:	68.1 m^3/hr (300 gpm)	68.1 m^3/hr (300 gpm)	205 m^3/hr (900 gpm)	181.7 m^3/hr (800 gpm)

Although availability and selection choices are improving, there are still only a few pump manufactures that are designing pumps for aquaculture applications, i.e. low head, high flow, energy efficient. Often pumps designed for a very different purpose find their way into aquaculture. For example, one of the authors has designed and built several small systems that use pumps originally specified for supplying water to the waterfall features in high-end decorative Koi ponds. As the flow increases, say beyond 114 m^3/hr (500 gpm), it is probably wise to seek professional help from pump manufactures, since there are many design details involved in the selection process to maximize performance. For small systems, under 91 m^3/hr (400 gpm), there are several manufactures of pumps that are cost effective for both freshwater and saltwater systems.

The flow requirements for Design Scenario One's Fingerling/Growout Pods are a center discharge total flow rate of 22.7 m^3/hr (100 gpm) and a sidewall discharge of 68.1 m^3/hr (300 gpm).

Figure 12.14 shows the pump performance curves for a Performance Pro Pumps ArtesianPro High Flow, 3450 RPM centrifugal pump, that one author has used in several system designs and completed facilities. Assuming that a propeller-washed bead filter is used for solids capture in the final design, the required pressure for operation is a maximum of 9.1 m (30 ft). Moving up from the bottom axis at 100 gpm, the first pump (AP1/2-HF) has too low of an operating pressure at this flow, the AP1-HF is too high, but the AP3/4-HF appears to be just right. In addition, if the discharge from the filter is completely shut off for some reason (deadheaded), the maximum pump pressure at zero flow is 41 ft (18 psi), which is less than the maximum pressure the filter is designed to handle (20 psi). The AP3/4-HF runs at 3450 RPM, 115/230 Volts, 60 HZ and draws 7.4 amps maximum at 230V.

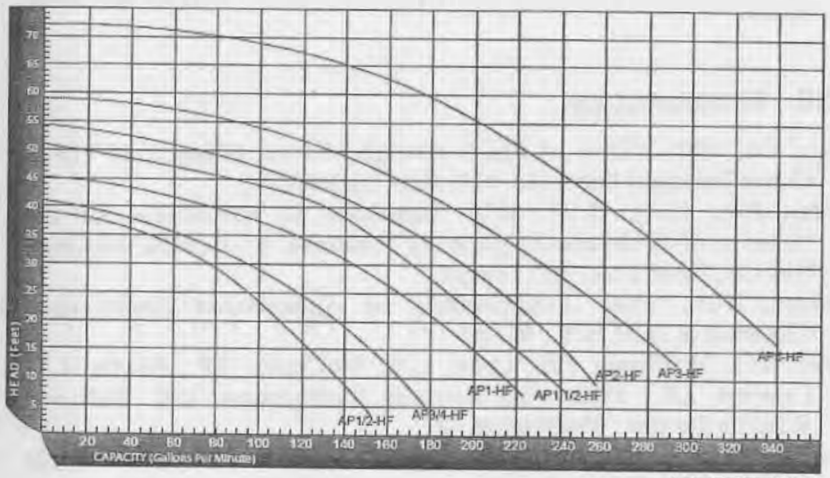


Figure 12.14 Pump performance curve for an ArtesianPro High Flow, 3450 RPM centrifugal pump.

Next assume that a Moving Bed Bioreactor has been chosen for biofiltration of the sidewall discharge of 68.1 m^3/hr (300 gpm). Since the pumping head for a MBBR is very small, most of the pressure generated by the pump will be used to re-inject the water into the tank to provide sufficient rotation to move solids to the center efficiently. Looking again at using an Artesian Pro pump, Fig. 12.14, one option would be to use the AP5-HF pump that would provide 300 gpm (68 m^3/hr) at about 30 ft of head. This is more pressure head then required and would also be

operated at the extreme of its performance curve, i.e. low efficiency. Another solution would be to use two pumps at 150 gpm (34 m^3/hr). Again the AP3/4-HF pump would work, providing 150 gpm (34 m^3/hr) at 15 ft (6.6 psi) of head. This works out extremely well since it means only one size pump is required for both solids capture and biofiltration, provides some flexibility in the flow rate to the MBBR and would require fewer backup pumps to be kept on hand, and might provide a significant cost discount due to volume purchase.

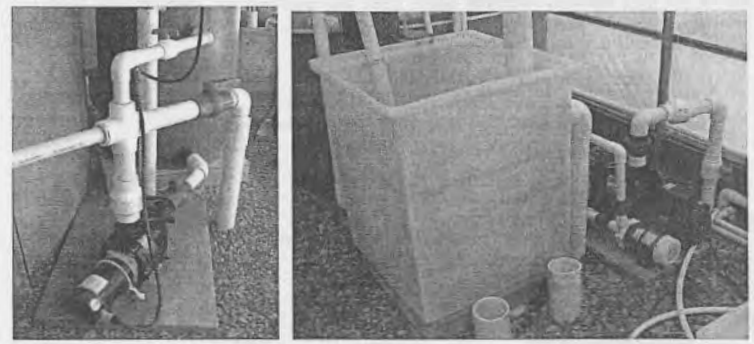


Figure 12.15 Two pump sumps.

A good rule of thumb is to design the pump sump to have a capacity of 3 to 5 minutes of continuous pumping and even more important, to have the top of the sump at the same level or higher than the normal water level in the tank. Then if the power goes out and the water drains back into the sump, it is not lost down the drain or on the floor! (This has happened in a saltwater research facility and flooded the lab!) Thus for the Design Scenario One with a center discharge total flow rate of 22.7 m^3/hr (100 gpm) suggests a pump sump of from 1.1 m^3 to 1.9 m^3 (300 to 500 gallons). And for the sidewall discharge of 68.1 m^3/hr (300 gpm), a sump tank volume from 3.4 m^3 to 5.6 m^3 (900 to 1500 gallons) is recommended.

Figure 12.15 shows two options for pump sumps used on a research marine system, a rectangular polyethylene tank and a larger cylindrical tank. Note the return line on the pump discharge back to the sump to allow the pump to work at its optimal discharge efficiency and the plastic pads under the pumps that help make for a clean, well laid out system. For this design example, since the sump is relatively large, a single rectangular fiberglass tank is chosen with a dividing partition between the solids capture side and the sidewall discharge side. The total volume

is between 1200 to 2000 gallons (4.5 to 7.6 m^3) or for this design a 4 ft x 4 ft x 8 ft (1.2 x 1.2 x 2.4 m) tank is used. Figure 12.16 shows a design sketch for a similar system for the production of grouper. The sump receives the discharge from both the center drains and the sidewall discharges. The center drains have an internal standpipe in the sump to maintain water level and allow flushing of the drainlines. This is accomplished by pulling the standpipes momentarily and allowing a large flow of water to scour the drains. The sidewall discharges have a standpipe next to the tanks, which also can be pulled to scour out these lines.

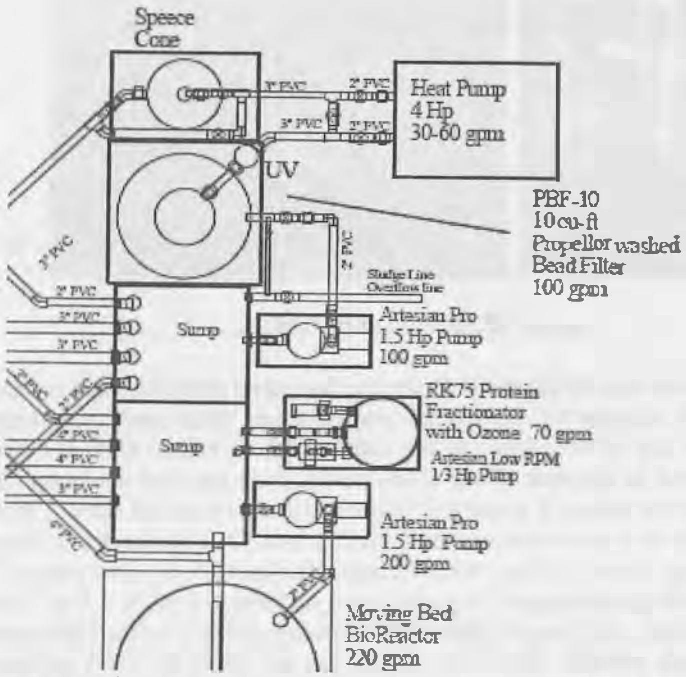


Figure 12.16 Design sketch (Grouper Fingerling/Growout production system) showing the sump for both the center drains and the sidewall discharges with two separate pumps for a PBF-10 (solids capture) and a MBBR (biofiltration).

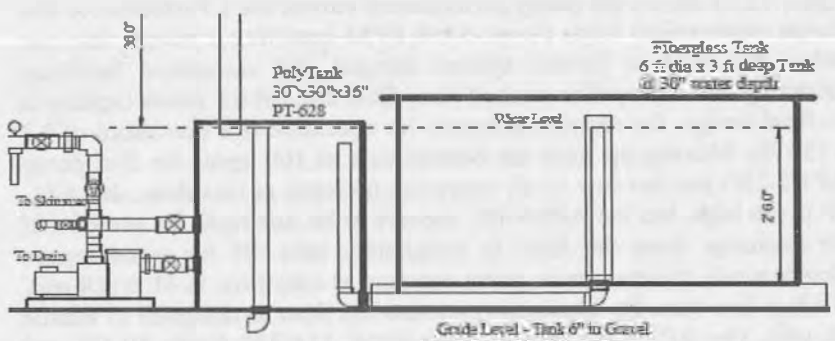


Figure 12.17 Design sketch (Grouper Fry production system) showing the sump for the center drains, inside standpipes and outside standpipe and pump details.

12.8 References

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List of Symbols

A	Cross-sectional area, ft^2 (m^2)
BHP	Brake horsepower, kW (hp)
C	Coefficient relating pressure to velocity,

dimensionless

$C_{H\text{-}W}$ Roughness constant used in Hazen-Williams Eq. 12.7
D Diameter of pipe, ft (m)
D_{inch} Diameter of pipe, inch
D_1 Diameter of upstream section, cm (in)
D_2 Diameter of throat or downstream section, cm (in)
E_{pump} Pump efficiency, %
f Friction factor (a function of the Reynolds number)
f_T Frictional loss coefficient that depends upon the size of the fitting
$FITTING_{constant}$ Frictional loss coefficient that is size independent of the fitting
g Acceleration of gravity, 32.2 ft/s^2 (9.81 m/s^2)
h Correction for channel kinetic energy, ft (m)
H_L Lost energy head due to friction, ft (m)
K Resistance coefficient for particular style of fitting
L Length of pipe, ft (m)
P Pressure of the fluid, psig (kPa)
P_1 Upstream pressure, psi (kPa)
P_2 Throat pressure, psi (kPa)
Q Mass flow, lb/s (kg/s)
TDH Total dynamic head that pump operates against, ft (m)
V Fluid velocity, ft/s (m/s)
V_1 Velocity in upstream section, ft/s (cm/s)
V_2 Velocity in throat of venturi, ft/s (cm/s)
Z Vertical distance from the reference plane, ft (m)
γ Specific weight of the fluid, lbs/ft^3 (N/m^3)[g]
ν Fluid kinematic viscosity, ft^2/s (m^2/s) (see Table 2.1)
ρ Density of the fluid, lb/ft^3 (kg/m^3) (see Table 2.1)
Ω_{sg} Specific gravity fluid, 1.00 for water, dimensionless

[g] It is important to note that here the "lbs" is in reference to force pounds (lbf), not to be confused with mass pounds (lbm). The official BG unit for mass it the slug. Note that one lbf = 1slug ·ft·s^{-2} and 32.174 lbm = 1 slug. The lbf is what we commonly measure as a pound, for example at the supermarket. Specific gravity is obtained by multiplying density by the gravitational constant, $\gamma=\rho g$. As density is a massed based unit, it is necessary to use the English unit of mass, as follows in this calculation of the specific gravity of water at 40°F:

$$1.940 \, {}^{slugs}\!/_{ft^3} \cdot 1 \, {}^{lbf \cdot s^2}\!/_{slug \cdot ft} \cdot 32.174 \, {}^{ft}\!/_{s^2} = 62.42 \, {}^{lbf}\!/_{ft^3}$$

CHAPTER 13

SYSTEM MONITORING AND CONTROL

13.0 INTRODUCTION

Intensive recirculation systems have the potential for a significant increase in production per volume of water, but at the increased risk of catastrophic loss due to equipment or management failures. In addition, the managers of these intensive production facilities need accurate, real-time information on systems status and performance, in order to maximize their production potential. At production densities approaching and even exceeding 120 kg/m^3 (1 lb/gal), failure of a circulation pump or aeration system can result in severe stress to the fish and probable catastrophic 100% losses within minutes. Expensive and sophisticated monitoring and control systems and components from other industries, such as the wastewater and petroleum industries, have been successfully modified for use in aquaculture. However, only a small fraction of their processing power is usually employed, due to aquaculture's relatively simple monitoring and control demands, i.e., digital inputs/outputs. Today, with the rapid decrease in costs for computers, software, and off-the-shelf monitoring hardware, systems of this type are within the reach of even small producers and are mandatory for large-scale production facilities.

Can you afford to purchase an automatic monitoring system? Just ask yourself what the value of your standing fish crop is and the losses to your business from an interrupted supply of fish to your customers. After doing this, there is only one obvious answer: minimally, you must have automatic and continuous monitoring of your most important water quality parameters, e.g., oxygen, and water levels, MINIMALLY! You can be in your facility and lose all your fish because of a drop in oxygen levels. You must do this!!

Before going any further, it should be emphasized that the most sophisticated (and normally, the most underappreciated and lowly paid), monitoring and alarm system is an attentive human operator. Experienced

staff can detect whether something is amiss the moment he or she steps into a facility, often just from the change in background noise. In the real world though, most facilities are not staffed 24 hours a day[a]. Moreover, the watery environment of aquaculture is totally different from what most "air-breathing" operators are accustomed to, requiring the need for automated monitoring of critical water quality parameters and system components.

13.1 PARAMETERS TO MONITOR

Murphy's Law simply states that: if anything can go wrong, it will (author's note: "and usually at 4:00 am on a Sunday morning"). Determining what can go wrong and generating a list of worst-case scenarios is a never-ending quest. From the authors' personal experiences, no matter how hard you try or how long your list is, there will always be a surprise in the near or far future and usually at the most inconvenient moment. Table 13.1 presents a short list of potential emergencies. It makes a good place to start, and then let your imaginations go wild, and assume that anything is possible, no matter how impossible it may seem!

Table 13.1 A Short List of Potential "Emergencies" in Intensive Recirculating Systems

Type / System	Causes
Beyond your control	Floods, tornadoes & hurricanes, wind, snow, ice, storms electrical outages, vandalism/theft
Staff errors	Operator "errors", overlooked maintenance causing failure of back-up systems or systems components, alarms deactivated.
Tank water level	Drain valve left open, standpipe fallen or removed, leak in system, broken drain line, overflowing tank.
Water flow	Valve shut or opened too far, pump failure, loss of suction head, intake screen plugged, pipe plugged, return pipe ruptures/breaks/glue failure
Water quality	Low dissolved oxygen, high CO_2, supersaturated water supply, high or low temperature, high ammonia, nitrite, or nitrate, low alkalinity.
Filters	Channeling/clogged filters, excessive head loss
Aeration system	Blower motor overheating because of excessive back-pressure, drive belt loose or broken, diffusers plugged or disconnected, leaks in supply lines.

[a] This choice depends upon production scale. Farms producing 250 tons or more per year will typically employ 24-hour coverage.

Keep in mind though, that it is also important during this initial design process not to go overboard in terms of technological complexity or in the sheer number of monitoring points and alarms. Sophisticated alarm systems are of little use, if the part-time help disarms them due to their unreliability and frequent false alarms. Has such a thing happened? Yes!

Table 13.2 Life Support Priorities in Intensive Recirculating Systems

High (fast response time – minutes)
electrical power
water level in tank
dissolved oxygen –aeration system/ oxygen system
Medium (moderate response time– hours)
temperature
carbon dioxide
pH
Low (normally slowly changing – days)
alkalinity
ammonia-nitrogen
nitrite-nitrogen
nitrate-nitrogen

When compiling a list of potential problem areas and water quality parameters, keep in mind the relative importance and the required response time each will require, Table 13.2. Life support priorities in aquaculture start with the presence of water being in the tank at the correct depth, followed immediately by adequate levels of dissolved oxygen. Then come the other water quality parameters, correct temperature, pH, and alkalinity, and finally, acceptable concentrations of ammonia-nitrogen, nitrite, nitrate, carbon dioxide, and suspended solids. At high stocking densities (greater than 40 kg/m^3, 1/3 lb/gal), dissolved oxygen requires the most rapid response time. If either water flow or aeration is interrupted for any number of reasons, low oxygen and the resulting stress can result in mortality within minutes; chronic or even short periods of a few hours of low oxygen can lead to disease problems. Thus in the design of intensive systems, a simple audible alarm in the office may not be adequate, or even a phone call if the manager lives 20 minutes or farther away. Therefore, in addition to the monitoring, some form of backup aeration must be provided for and automatically engaged

to insure survival of the fish. Except for dissolved oxygen, most of the other water quality parameters change relatively slowly and can take hours or days to reach levels of concern. This allows more time to discover and analyze the problem and take the necessary steps to correct them.

At low stocking densities (less than 40 kg/m^3, 1/3 lb/gal), basic parameters to be monitored include system electrical power, tank water level (high and low), aeration system pressure, and water flow through the filters and tank. All of these parameters can be monitored by simple digital sensors, i.e., either on or off. Analog sensors, such as dissolved oxygen levels and pH monitors, are more expensive to buy, install, and operate. Continuous dissolved oxygen monitoring is crucial at high stocking densities or whenever oxygen is used. For all these critical parameters, it is equally important *where* you monitor parameters, as *what* you monitor. Common sense should be the guide in this aspect of monitoring. For example, it is of little value to monitor the flow from a pump into a tank, if the tank drain line is left open, and all the water is flowing out as fast as it is flowing in. What is important to the fish is the tank water level. Similarly, there is no advantage in monitoring the power to a pump, if the discharge valve is shut or the motor thermal-overload switch has turned the pump motor off. The critical parameter to monitor is whether there is flow from the pump. Finally measuring air pressure next to the aerator is of little help, if there is a major leak at the far end of the distribution system, resulting in low air pressure for the last tanks on the line. The aeration pressure needs to be monitored at the farthest point in the system or at several different points. Table 13.3 lists some of the important systems and parameters that need to be monitored in intensive recirculation systems.

Sensors in aquaculture can be roughly divided into two major types: digital (on/off signals) such as water level, aeration pressure and water flow switches and analog (continuous output) such as dissolved oxygen, temperature, pH, conductivity, and ammonia-nitrogen probes. In addition, most analog probes require some additional hardware or controllers to convert the probe's output to a usable signal, provide a digital display, and allow for calibration and zeroing. Thus the higher cost for these types of measurements, compared to simple switch closures. What follows is a short review of important monitoring parameters.

Table 13.3 Important Systems or Parameters to Monitor

Electrical power	Single and three phase supply, individual systems on GFIC's
Water level	Culture tank (high/low), supply sumps to pumps (high/low), chemical storage tanks, head tanks/ reservoirs (high/low), filters (high/low)
Aeration system	Air/oxygen pressure (high/low)
Water flow	Pumps, culture tanks, submerged filters, in-line heaters
Temperature	Culture tanks (high/low), heating/cooling systems (high/low)
Security	High temperature/smoke sensors, intruder alarm

Data that is not recorded is useless if one tries to trouble shoot a problem or to investigate trends in performance. A simple data log sheet to record both fish performance and water quality chemistry is an absolute must (see Figs. 13.1 and 13.2). This daily data should then be transferred to a master spreadsheet for plotting results. Post these results so your staff can see them and also show expected biological performance for the fish as target goals (e.g., current weight and running feed conversion).

Water Quality Data Sheet:

Tank Identifier ____________________

Date/Time:	DO (mg/L)	Temp (°C)	Salinity (ppt)	pH	TAN (mg/L)	NO_2-N (mg/L)	NO_3-N (mg/L)	Alkalinity (mg/L)	Turb (NTU)	Feed (kg)	Data Logger Info	Daily Notes
5/1												
5/2												
5/3												
5/4												
5/5												
5/6												
5/7												
5/8												
5/9												
5/10												
5/11												
5/12												
5/13												
Add days to finish month												

Figure 13.1 Sample recording chart for daily water chemistry parameters.

Feed Data Sheet:

Date:	Time:	Tank #1 (gms)	Tank#2 (gms)	Tank#3 (gms)	Notes
5/1					
5/2					
5/3					
5/4					
5/5					
5/6					
5/7					
Add days to finish month					

Figure 13.2 Sample recording chart for daily feedings; one chart for each tank can be used with chart at the tank location.

ELECTRIC POWER

Power failure is probably the most common emergency and the one most easily monitored. Monitoring power is especially important when systems such as filters or supply pumps are located some distance from the main building. Three-phase power can be especially confusing, because if only one phase is down, it is possible to lose power to some systems, but not all. Murphy's Law assures that the monitoring system will not be on the one phase that goes down. In addition, when power is lost on only one phase, severe damage can occur to three-phase motors and pumps, if not properly protected. One often overlooked result of power outage is the loss of lights, which means that either numerous flashlights need to be maintained in good working order or back-up emergency lighting provided. When the power does go out, back-up generators suddenly become worth their weight in gold, as long as they have been properly maintained, regularly tested, have sufficient fuel and come on line when needed.

WATER LEVEL

Water level is probably the easiest and most inexpensive parameter to measure, and should be monitored for both high and low levels in each production tank. High/low water level sensors will detect plugged drain lines, fallen standpipes, and make-up water hoses left on or drain lines accidentally left open. Other locations to monitor include the intake side of pumps in wells or sumps. These should provide for automatic shutdown of the pumps to prevent their damage in the case of low water levels. Supply reservoirs or head tanks need to be monitored for both high and low levels. High levels can indicate unusual change in normal water demands, due to clogged pipes or valves accidentally turned off. Low levels can be caused by pump or water supply failure. If immersion heaters are used, low level monitoring should be designed to turn them off, to prevent overheating and burning out of the heaters and melting PVC pipes. Water level alarms should be set so that normal operating transient do not activate an alarm. This can be accomplished either by setting the levels optimistically or by allowing some time delay before an alarm is activated after a sensor is triggered. Level sensors should be protected, so that active fish do not accidentally trigger them or in some cases chew on them.

AERATION SYSTEM PRESSURE

The aeration system is one of the most critical systems in any intensive recirculating aquaculture system. Response time to a detected failure is very short, and both monitoring and backup systems are important. Low pressure in the system may mean a ruptured airline, open or jammed pressure relief valve, disconnected diffusers, or blower failure. Although not monitored as often, excessive high pressure could indicate blocked supply lines, valves turned off or clogged diffusers.

WATER FLOW

In some cases, the actual measurement of flow rate is important, such as when needed for the proper operation of chemical injection systems, for dechlorination, or for monitoring system performance. Normally however, simply monitoring whether water is actually flowing (flow/no-flow) with a digital sensor is adequate. One example of systems that these sensors protect are in-line heaters that require continuous water flow to prevent overheating and meltdown. Another example is submerged biological filters, where anaerobic condition due to pump failure can damage the nitrifying bacteria.

DISSOLVED OXYGEN

Dissolved oxygen (DO) is one of the more expensive and difficult parameter to monitor continuously. Thus, the decision whether or not DO should be continuously monitored is dependent on the overall economics of the system, stocking density, and degree of risk a manager is willing

to accept. Normally, the actual value of DO (mg/L) is not needed, just whether it is above or below a given set point. However, to provide a simple digital signal, both an expensive probe and a sophisticated hardware interface are required. The availability and costs of both oxygen probes and interface hardware has dramatically decreased in the last few years, but still remain high for many aquaculture operations. But if you use pure oxygen, ***you must monitor DO continuously***.

Temperature

The continuous and precise monitoring of temperature in production tanks is important to optimize production, reduce stress, and minimize risk of disease. Systems should be monitored for both excessively high and low temperatures; keeping in mind the two extremes are not equal. While low temperatures may reduce growth, excessively high temperatures may yield a huge tank of fish soup and a new career path. Since most temperature controllers are cyclic in nature (either on or off), temperature alarm limits should not be set too close together, to prevent unnecessary alarms due to short term transients.

Other Water Quality Parameters

Other water quality parameters, pH, ammonia-nitrogen, nitrite, nitrate, alkalinity, and carbon dioxide change relatively slowly in comparison to dissolved oxygen. Although relatively expensive individual probes and automated systems are available to monitor these parameters, the most cost-effective method is daily or weekly manually monitoring with simple chemical analysis techniques.

Physical Plant Security

Intrusion alarms, smoke, and high temperature sensors (fire) are readily available and commonly used to protect against fire, theft, and vandalism. Often existing alarm systems can be connected to the proposed monitoring system.

13.2 Monitoring Sensors and Equipment Options

Over the past few years, the cost of computer hardware and software has dramatically decreased, while the processing power and computer programming sophistication has greatly increased. Sensor technology has become more reliable and sophisticated with such innovations as embedded microchips in the sensors that provide signal processing and

linearization. Many analog meters come with RS-232/ RS-485, Ethernet and web capability built-in. It is even possible to obtain wireless monitors and sensors that allow monitoring of remote systems without the high cost of running dedicated cables.

In the past, a large number of sensors and monitoring systems components were adopted from the wastewater treatment and chemical and petroleum industries. In many cases, the sophistication and corresponding expense of these types of monitoring and control equipment was not necessarily required in aquaculture facilities. More recently, there have been several manufacturers of these types of monitoring and control systems that have developed a product line especially designed for aquaculture.

The following description of sensors represents the simplest and most cost-effective solutions to monitoring each individual parameter. These suggestions are not the only solutions available, however. For each of these parameters, there is a multitude of potential solutions, some of which are more expensive, more accurate, more reliable, more precise, with better interface capabilities, or simply more readily available. There is no simple right or wrong answer, and this is where the system engineering and design requirements come to play. Keep in mind, that for any monitoring system, its overall reliability is determined by the most unreliable part, i.e., the weakest link.

Water Level – Float Switches

Water level is probably the easiest and most inexpensive parameter to measure. The basic float switch is designed to monitor a single, discrete, preset liquid level, Fig. 13.3. Simple float switches are constructed with a float containing a small magnet, which moves with the water level and actuates a hermetically sealed reed switch within the stem or body of the float switch. The rugged construction of this design provides for long and trouble free service with minimum maintenance requirements. Several different designs are available for mounting either vertically or horizontally in the tank or sump. Two float switches can be wired in series to monitor both high and low levels in a tank, Fig. 13.3. Although most float switches are designed to handle 110 VAC at small currents, they should be powered by low voltages, i.e., 24 VAC or 12 VDC, to minimize danger to personnel and fish. Float switches are simple, foolproof, and inexpensive.

Other options for monitoring water level include optical liquid-sensing sensors that use an internal infrared circuit and the light refracting properties of water. Non-contacting ultrasonic level sensors measure the time required for the ultrasonic pulse to travel to the water

surface and return. Conductivity level switches operate by detecting a small electric current between a single electrode probe and a grounded metal tank or between two electrodes. Finally, pressure-sensing systems use a pressure transducer to measure the pressure required to bubble air through an immersed pipe in the water column. Each of these has their application in specific design situations.

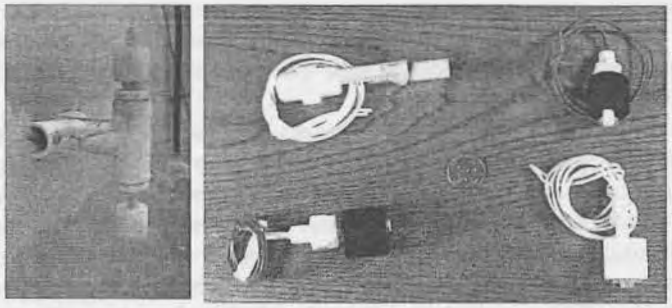


Figure 13.3 Level sensors or float switches for monitoring tank water levels.

AERATION PRESSURE

Pressure is defined as a force per unit area, which is to be used to produce a deflection, distortion, or some other physical change in a sensor. A pressure control switch uses this deflection to trip an electrical switch at a preset pressure setting. Low and high pressure switches are available in a wide variety of configurations and price scales, from numerous manufacturers, Fig. 13.4.

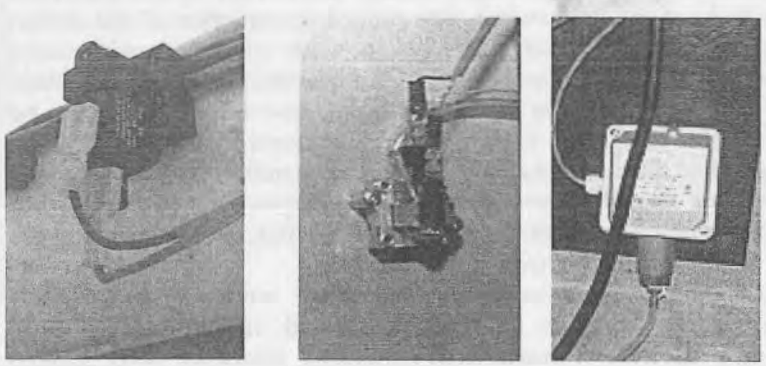


Figure 13.4 Pressure switches for monitoring aeration or water pressure.

WATER FLOW - DRAG DISCS, PADDLE AND VANE FLOW SWITCHES

Drag discs, paddle and vane flow switches are all designed to monitor flow/no-flow or low flow conditions. Each operates on the drag force of the moving water against a small disk, paddle, or vane in its path, which in turn controls a small micro-switch. They are available in a wide range of flow rates and pipe sizes. Normally drag discs and paddles are installed using a Tee fitting and vane types are installed in-line, Fig. 13.5.

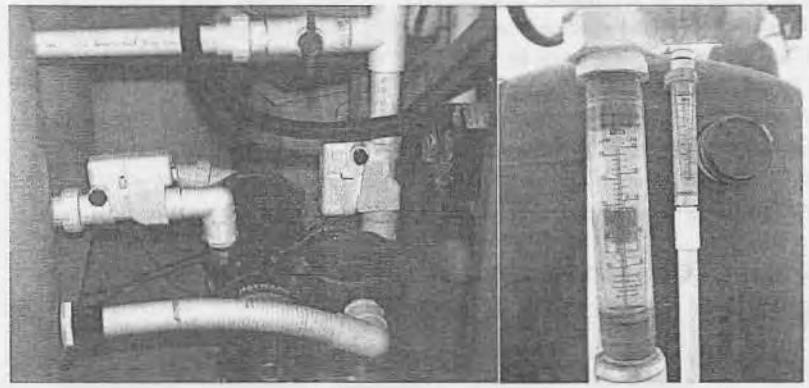
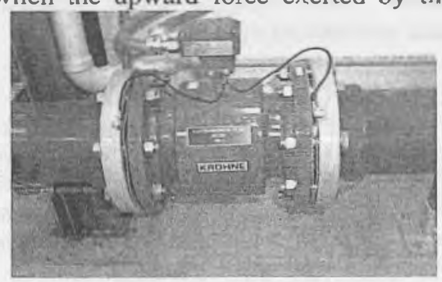


Figure 13.5 In-line paddle flow monitor and rotameter flow meters for monitoring water flow rate and air flow rate.

Other options for monitoring flow are rotameters. As the water flows through the rotameter, it raises a float in a tapered tube, increasing the area for passage of the fluid. The greater the flow, the higher the float raises in the tube, which is directly proportional to the flow rate. The float reaches a stable position, when the upward force exerted by the flowing water is equal to the downward force exerted by the weight of the float. These flow meters can be used to monitor flow by mounting a proximity switch externally, which is switched at a predetermined flow rate by a small magnet in the float. A second more

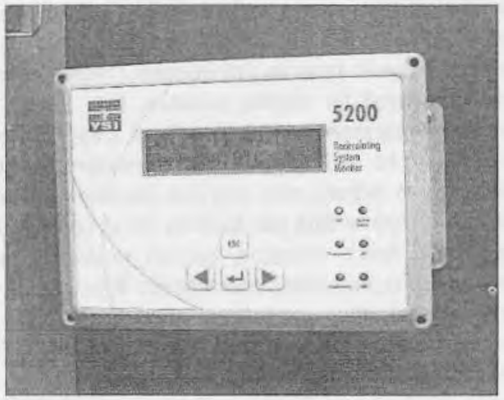


expensive option is to use a turbine or paddlewheel flow meter. The flowing water turns a small turbine blade or paddlewheel, which

generates an electrical pulse. This pulse is sent to the appropriate hardware, where the flow rate or the total flow can be displayed, and alarm conditions set and low/high flow alarm relays activated.

DISSOLVED OXYGEN

Over the past few years, a number of dissolved oxygen probes and analyzers designed specifically for the aquaculture industry have become available. Most of these are microprocessor-based instruments capable of measuring levels of dissolved oxygen up to 100 PPM, important for monitoring oxygen injection systems. Standard recorder outputs (0–5 VDC) are built-in and many include 4–20 mA current loop outputs. Several models also provide Ethernet and serial outputs (RS-232, or RS-485) for direct interfacing with microcomputers and local area networks (LAN's). And most recently, several manufactures have added wireless communications to a host computer in the air-conditioned office from one or multiple monitoring systems. These also include high/low set point control relays for automatically controlling external devices such as aerators, pumps, valves, or other alarm monitoring equipment. As an example, YSI has designed two monitoring and control systems specifically for aquaculture, the Model 5200A which can continuously monitor and log DO, pH, conductivity, ORP, salinity, and temperature and the Model 5400 which can continuously monitor and log four DO probes and four additional inputs such as temperature, pH, and ORP. Both models include sophisticated alarming modes that include local alarms and responses such activation of backup oxygen, and with a modem or software, up to 3 emails to alert management. The Model 5400 comes with 8 relays available for control outputs, while the Model 5200A comes with 4 internal relays with switching capabilities. The relays are accessible through terminal blocks on the I/O panel inside the enclosure. The terminal blocks provide Normally Open, Normally Closed, and Common Connections to the relays.



Although the initial investment in this equipment can be high, the cost must be weighed against the potential loss and poor growth due to

low dissolved oxygen levels. Dissolved oxygen meters designed specifically for aquaculture are available from Point Four Systems, Inc. (www.pointfour.com), YSI, Inc. (www.ysi.com) and Campbell Scientific (www.campbellsci.com), Fig. 13.6.

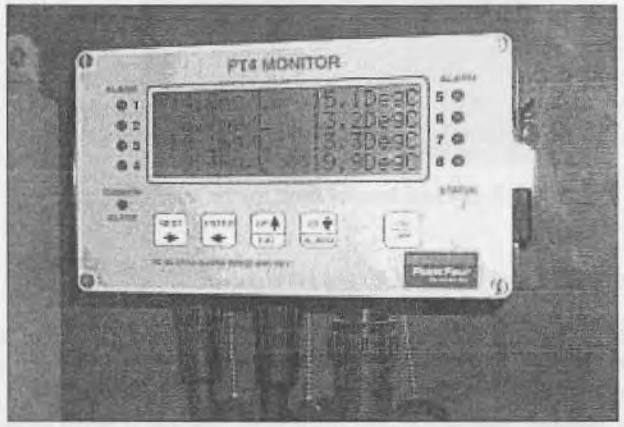


Figure 13.6 Dissolved oxygen meter from Point Four (www.pointfour.com).

OTHER WATER QUALITY PARAMETERS

Temperature, pH, and other water quality parameters usually have a relatively long response time, when compared to dissolved oxygen or the effects of pump or aeration failures. Thus, these and other slowly changing parameters are probably best measured as part of a regular water quality monitoring program, using bench-top laboratory equipment. Although equipment exists for continuous monitoring of pH and temperature, Fig. 13.7, except for research purposes, it is not normally required in production aquaculture. On-line ammonia monitoring is possible, but in practice, it is very expensive and difficult to accomplish.

Figure 13.7 pH and temperature meter from GLI.

13.3 AUTOMATIC PHONE DIALERS

The final step in the development of the monitoring/control systems is to bring each of the potentially catastrophic alarms to the attention of the manager and staff, especially when they are home sleeping. A very inexpensive, simple, and versatile monitoring system can be constructed around readily available automatic telephone dialers/alarm systems. These units are readily available from several manufactures for a wide range of inputs, sophistication, and costs. One such unit, the Sensaphone® Model 400 (Phonetics, Inc., Aston, PA, (www.sensaphone.com) has been used in several research and production facilities by the authors with excellent results. One unusual application was to monitor water temperature and levels in a lobster holding facility for a large restaurant chain in New Orleans.

The Sensaphone unit automatically monitors the following conditions:

- AC electric power – power failure
- four digital alert inputs or temperature sensors
- temperature – reports actual temperature and checks for high or low limits
- high sound level – fire/smoke alarm, intruder alarms, "unauthorized parties"
- battery status – condition of its battery back-up

All monitoring is continuous and when an alarm condition occurs, the unit announces the alarm status locally and can activate an alarm relay to set off a local light or siren. If no response is received, it then

sequentially dials up to four to eight user-programmed telephone numbers (including pagers) with an alarm message. It will replay a customized voice phrase in your own voice to describe each alarm condition existing and wait for an acknowledging telephone call or coded response. It will continue dialing-out until its message is properly acknowledged. In addition, it is also possible to call in, listen to a status report on the monitored conditions, and hear the background sounds through a built in microphone. For most small systems, this would provide all the necessary digital inputs for monitoring tank water level (high/low), aeration systems pressure, water flow, and sump water level or if desired, system water temperature. If additional monitoring points are required, the Model 800 allows up to eight digital inputs or temperature sensors.

A BASIC MONITORING SYSTEM

A *basic monitoring system* was designed (and is used by one of the authors) for a low-density (less than 40 kg/m^3 or 0.33 lb/gal) recirculating systems, with aeration only and moderate feed rates, such as broodstock holding tanks, isolation/quarantine tanks, or educational systems. Basic system parameters (water level, air pressure, water flow) are monitored by digital sensors, i.e., either on or off. Analog sensors, such as temperature and dissolved oxygen levels, are more difficult and expensive to utilize and are important only at much higher stocking densities or where pure oxygen aeration is required. Basic parameters to be monitored at this production level include electricity, tank water level (high and low), aeration system pressure, and water flow. The actual number of subsystems monitored, depends on the specifics of the system design and the operating conditions. In most cases, only a few monitoring points should be necessary. Parameters monitored and examples of the sensors used include:

- **System electrical power**: monitored directly using the Sensaphone (Fig. 13.8) or indirectly due to loss of other subsystems (pump flow, aeration, etc).
- **Tank water level (high/low):** Aquatic Eco-Systems, Liquid Level Switch ST3M, or Grainger Liquid Level Switch 2A554, wired in series.
- **Aeration system pressure**: Aquatic Eco-Systems, Pressure Switch B601.
- **Flow-sensing switch**: Aquatic Eco-Systems, Flow Switch ST9.
- **Telephone dialer**: Sensaphone® Model 400

Each of the sensors is wired directly into a Sensaphone input, with the two float switches wired in series (Fig. 13.3) to monitor both high and low water level. The fourth input on the Sensaphone could be used to monitor either temperature of the water or an additional alarm. With this system design, a single tank or perhaps several could easily be monitored for the basic system parameters: water level, flow, aeration, and electricity.

Figure 13.8 The Sensaphone for continuous monitoring and phone dialer (older model).

13.4 Backups Systems are not an Option!

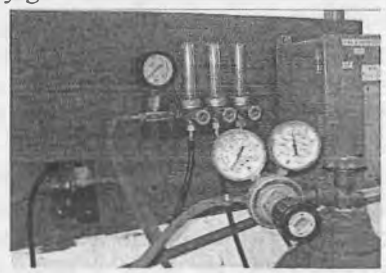
(Reprinted from a Global Aquaculture Advocate article, March 2010)

It is 5:14 in the morning when my cell phone rings! It is the dreaded phone call, the primal fear of any aquaculture manager: "This is Sensaphone number 555-1212, The electricity is off, The electricity is off!" I am dressed and out the door in less than 10 minutes. Breaking every speed limit on the way I burst through the greenhouse doors, hoping that the fish are not floating on the surface for lack of oxygen. Without electricity, the regenerative blowers are off, the circulation pumps are off, and the overhead lights are off! Luckily, the flashlight is where it is supposed to be and it even works! The question was: were the fish still alive after 20 minutes without electricity or not? Much to my delight the answer was not only "yes", but the dissolved oxygen in the

tanks was actually higher than normal and the fish, although spooked due to lack of circulation, were fine. How was this possible you ask? It was because of a simple backup system that turned on an oxygen diffuser in each tank as soon as the electricity went off and stayed on until the electricity went back on again. Lesson learned: for less than $200 in equipment, the backup oxygen system not only worked when needed, but saved the day!

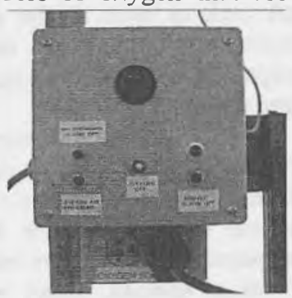
During our visits to numerous research and commercial aquaculture facilities, we are always amazed to find either no backup systems or systems that have not yet been installed or operational. Yet, the very survival of the research programs, the endangered species, or the commercial venture rests on the successful growout and high survival of the target species. Why, we repeatedly ask, are the monitoring and backup systems not installed? The answer is invariably that "we haven't gotten around to it" or "the money ran out before we could get to them" or the "specialist' left and no one knows how to turn it on". As a result, we all read several times a year about the loss of all the fish in a facility due to something as simple as a loss of power. Or if the fish are not completely lost, then they are stressed so much that after several weeks they suffer from stress induced diseases.

Backup systems do not have to be sophisticated, complicated, or expensive. In fact, the simpler they are, the more likely they are to work (and to be used!). For intensive recirculating aquaculture systems, just like for NASA, failure is not an option. Since the loss of power is one of the most common emergency situations, we have always had at least two immediate responses when the electricity goes off. The first is immediate notification of at least two people through the use a of inexpensive, simple, off-the-shelf automated phone dialer that can be purchased from several manufacturers. At a minimum, these will dial out to three or four phone numbers and announce the emergency condition so that someone can respond as quickly as possible. In addition, they usually have several on/off switch inputs that will activate an alarm locally, call someone, and announce the alarm condition.



The second response is to minimize the impact of the emergency situation on the aquaculture system. A backup generator that automatically starts when the power goes off and has a power transfer switch is the best solution to power outages, but often too expensive to purchase and requires routine maintenance and upkeep. For a quick

response to a power outage for small scale operations (large scale will have 24 hour people on site in addition to the monitoring system), one option is to have several high pressure cylinders of oxygen that are connected to an inexpensive Normally Open (NO) solenoid valve that is simply plugged into a wall outlet. When there is power, the solenoid is energized and closed, when the power goes out, the solenoid opens and oxygen flows. An oxygen manifold then directs oxygen to each aquaculture tank and to an oxygen diffuser. The system is straightforward to build and install. In addition, this set-up can be used when sorting or moving fish to add a little extra dissolved oxygen to minimize stress. But like Murphy said: If it can go wrong it will! I did find a loop-hole in the system when hurricane Gustav struck New Orleans last year. The problem was that the electricity stayed on, but the aeration system manifold was broken. And even though I received a call reporting the low air pressure, I was several hundred miles away and unable to respond. The solution to this problem was to add an air pressure switch and a relay to the oxygen backup system, so that if the aeration pressure failed, the oxygen solenoid opened and oxygen flowed to the tanks.



The other major emergency situation that we have experienced over the years is loss of water. This may be hard to imagine, but in one case a pipe broke, another time a rubber coupling came lose, a third time a hose fell into the tank and siphoned out the water, one time a drain line was left open. I once left a hose running in a tank over the weekend and all the fish were on the floor on Monday morning. Water level is one of the easiest parameters to monitor using a float switch. We monitor both low level and high water level with two float switches wired in series, so that if one is activated a local audible alarm goes off. This allows us to maintain water level in the tanks within a very narrow range. In addition, a second low level float switch is installed at a depth of 30 to 40 cm and is connected to the phone dialer. Thus small changes in water level are reported locally for quick response by staff and any significant drop in water level is phoned to the staff. As an added feature, these float switches can act as a safety shut off for submerged heaters in a tank in the event of a dramatic drop in tank water level.

So, it was 5:20 AM, the dissolved oxygen levels are fine, but there is still no power and the transformer on the pole down the street is still arcing and flashing. We have several portable backup generators on site that are easily rolled next to the greenhouse for restoring aeration. Of course, as Murphy will remind you, something always goes wrong. Even though we try to test these generators regularly, I was unable to start the primary generator. But like all good backup plans, there should always be a backup to the backup and our second backup generator started without any problem. A couple of extension cords and within a very short time, aeration and circulation were restored in the research greenhouse. The only thing overlooked was a provision for emergency lighting so that we could see what we were doing. That was rectified for future events.

The moral of this tale is to expect the worst, plan for it, but above all do not think that backup systems are too expensive or too much of a bother to install. Before you stock any fish, you should have a minimum of backup oxygen and portable backup generators available for emergency situations. You may not have to plan for hurricanes, but even a burned out transformer could lead to an unplanned career change.

13.5 COMPUTER BASED SYSTEMS

Once it becomes necessary to monitor analog inputs, such as dissolved oxygen, pH, temperature or other output analog signals for control purposes, some form of computer-based system must be employed. With this added capability though, comes the cost of additional requirements for calibration of the probes and sensors and maintenance of the overall system. Until recently, the utilization of computer control and monitoring systems in aquaculture has been limited, with only a few custom-designed systems for research or large commercial operations. The vast majority of small producers has had neither the expertise nor the resources to custom-design and installs systems. However, in the past years, there has been a revolution in low-cost, high performance monitoring systems and intuitive and relatively low cost process control software.

Supervisory control and data acquisition (SCADA) is the term used in industrial automation that refers to automated data collection usually by remote units and its display on a centrally located personal computer. The human machine interface (HMI) is the interface by which an operator interacts with the remote devices, i.e., pH, DO, temperature measurements. The HMI allow an operator to adjust set points, configure remote units, respond, and acknowledge alarms. In addition, system performance data can be recorded for future analysis and solutions to recurring and one-time problems. A data acquisition system has a variety of input/output (I/O) ports that provide connection between the sensors and the computer. Table 13.4 shows the four of the most commonly

used: digital input (DI), digital output (DO), analog input (AI), and analog output (A●). Determining the type of I/O required by sensor is the first step to matching the sensor and the required I/O.

Table 13.4 I/O Ports Connecting the Computer to Sensors

I/● Type	Applicable sensors or control equipment
Analog input	DO, temperature, pH, flow sensors (voltage, current)
Analog output	Proportional control of chemical feeds, pump speeds
Digital input	Level switches, valve status, counters
Digital output	Activate relays controlling feed, oxygen, pumps

Computer control and monitoring systems in aquaculture can be separated into two design strategies:

1) stand-alone programmable logic controller (PLC) systems, or closed loop controllers,
2) centralized microcomputer based system.

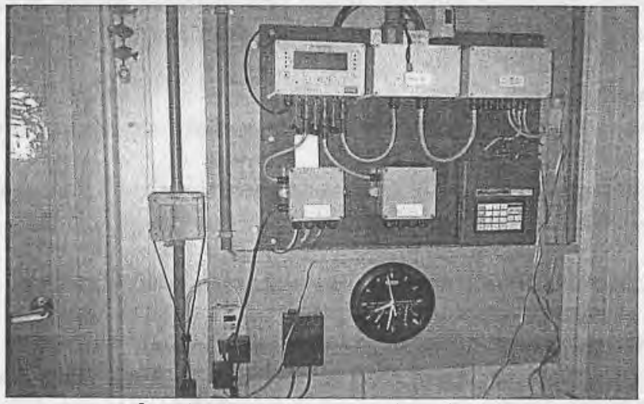


Figure 13.9 Stand-alone system based on the Point Four PT4 meter with digital input monitoring (www.pointfour.com).

The first design strategy can also be expanded to a distributed process control system, where each of the stand-alone monitor/control units relays data to a central supervisory microcomputer, Figs. 13.9 and 13.1●. Examples of the stand-alone closed loop controllers are dissolved

oxygen analyzers (YSI, Royce, and Point Four Systems, Inc.) and temperature controllers. Each of these closed-loop controllers is normally equipped with both control relays and high/low alarm relays. In general, these units have limited data display capabilities and normally do not store data, but are equipped to transmit the data to a central, supervisory microcomputer. With stand-alone systems, individual sensors are easy to service and calibrate, since each has its own hardware and display unit. In addition, monitoring and control is performed at the lowest system level, which provides a high degree of overall system stability and robustness. If a failure occurs in the supervisory microcomputer, the stand-alone units will continue to monitor and control critical processes. If a failure occurs in the stand-alone system, the supervisory microcomputer can by comparing to previous measurement or measurements from other sensors to detect the abnormal conditions and alert the operator.

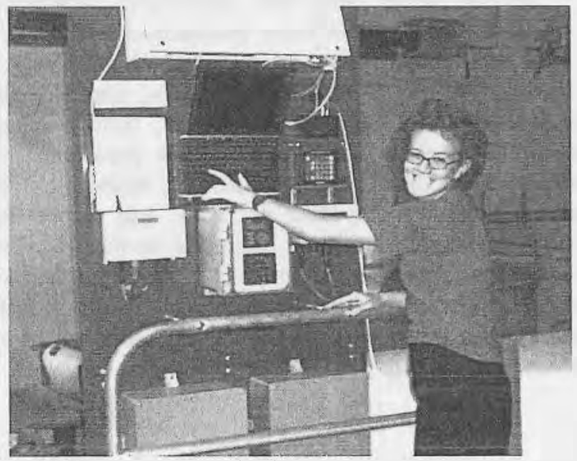


Figure 13.10 Control monitoring system at Freshwater Institute. (Author's daughter, Margaret Timmons, inspecting system.)

The second design strategy utilizes commercially available data acquisition boards that are either located in existing expansion slots in the computer or communicated to it via a serial interface link or dedicated data acquisition systems such as the Campbell Measurement and Control System. There is a wide selection of data acquisition boards available over a wide range of c●st, performance and sophistication, including analog to digital (A/D) cards for monitoring voltages or

currents from sensors, digital to analog (D/A) cards for outputting analog control voltages, and input/output cards (I/O) for monitoring and outputting digital control signals. They are easy to use, "just-plug-in", and come with a set of standard drivers and application software programs. Many types of sensors can be connected directly to these boards and most meters usually have some form of recorder output (0–5 V or 4–20 mA). In contrast to the distributed system, the microcomputer operates as the primary controller, monitoring, recording data, and controlling alarm functions. These systems are not as inherently reliable as the distributed systems, but overall systems cost is less, since they are based on fewer and less expensive components.

13.6 Design Example – Monitoring

The monitoring systems for the Omega fish production system starts with the previously described ***basic monitoring system*** since production is at a low-density (less than 40 kg/m^3 or 0.33 lb/gal), requiring aeration only in the fry/quarantine tanks, and the fingerling systems. Basic system parameters (level, pressure, flow) are monitored by digital sensors, i.e., either on or off. Basic parameters that are continuously monitored include facility electricity, individual tank water level (high and low), aeration system pressure, and water flow. The actual number of subsystems monitored, depends on the specifics of the system design and the operating conditions. A Sensaphone 400 or equivalent is used to phone staff in emergencies situations and of course, a backup oxygen system is installed in the case of aeration or electrical power failure before the fish are put into the tanks!!!

13.7 System Design and Maintenance

Listed below are some general suggestions about overall system design and maintenance:

System Design:

- Choose sensors carefully, use the fewest possible, label everything, and include expansion capability in all components.
- Aquaculture facilities are now included under the National Electric Code; it may not be of concern to you, but it is to your insurance agent.
- Mount sensors and equipment where they are visible and easily accessible for service and calibration.
- Remember that water and electricity make for a fatal combination, so use low signal voltages (5 VDC, 12 VDC or 24 VDC or AC) to protect you and the fish.
- Clearly label the sensor's armed and unarmed modes preferably with LED's at each station to show sensor status.

System Maintenance:

- Have a well prepared maintenance manual accessible to the staff.
- Maintain a weekly/monthly/yearly maintenance scheduling plan and files of major service records and equipment manuals.
- Maintain daily/weekly/monthly instrument check lists.
- Perform regular (and some unannounced) system checks, including triggering of each sensor and checking operation of the automatic backup systems and phone dialer.
- Provide staff training to handle routine alarms.
- Ensure staff familiarization with the complete operating system, including water supply, aeration, and emergency backup systems.

13.8 CONSTRUCTION HINTS

Probably the most important rule during design and construction is to *keep it simple stupid*, known as the "KISS" principle. The other rule is always assume that someone else will have to repair it, thus complete design notes, wiring diagrams, and labels are important. If you change a configuration of a monitoring system, update the documentation, and date the update. In addition, system components should be readily available from local or reliable sources. A "one-of-its-kind" is just that, and will soon become extinct. While designing and constructing, plan for expansion and leave room for additional systems or more "bells & whistles".

Monitoring and Control equipment should be mounted and operated in a clean, dry, and safe environment. Provide adequate space around and in front of the equipment for easy access and future expansion. Do not place equipment were it will be subject to shock and vibration, dirt, dust or moisture. As much as possible, all materials used for system housing and hardware should be PVC, fiberglass or stainless steel to minimize corrosion. Water-resistant PVC junction boxes and fiberglass electrical cabinets, NEMA-4 enclosure, are corrosion resistant and easy to drill holes into and are ideal housing to protect the electrical components, Fig. 13.11. Include several vent holes to minimize heat build up in the cabinets. Do not install the system near motor starters, contactors, or relays that switch inductive loads, i.e., motors! These devices generate large electromagnetic fields that can cause communications and system errors. If unavoidable, install the system in separate, grounded, steel enclosure to the shield from electrical interferences.

Figure 13.11 Fiberglass electrical cabinet to protect Sensaphone and relays.

All external sensors should be low voltage, i.e., 24 VAC or 12 VDC, ON/OFF, to minimize danger to operators and fish. Crimp style quick disconnect tab connectors on switches allow for easy construction and later modification. Solder joints should be covered with shrink-wrap tubing whenever possible. When buying individual components, look for extra options that may be useful in the future, such as extra alarm relays, voltage or current outputs and computer interfacing capabilities.

One simple trick to minimize the effects of aquaculture's harsh environment is to pressurize the control system housing using the aeration air supply. In this manner, relatively dry air is forced into the housing, preventing the high humidity and salt air from getting in. Alternatively, the step-down transformer in many of the systems provides a source of heat, thus preventing condensation from occurring.

WIRING CONSIDERATIONS

(adapted from Phonetics, Inc., Sensaphone SCADA 3000 manual Version 2.0)

Most sensors can be connected using inexpensive 2-conductor twisted-pair cable as small as #24 AWG (up to 700 ft, 210 m). For wiring distances up to 1500 ft (450 m) use #22 AWG and for distances up to 2500 ft (760 m), #20 AWG. If the senor is located far from the monitoring unit or if the cable is running in an electrically noisy environment, twisted pair shielded cable should be used. This will shield the signal from electrical interference and prevent false readings and possible damage to the monitoring system. When using shielded cable, you only need to connect the shield to the earth ground at the monitoring system, not at both ends. The guidelines below are designed to minimize electrical noise coupling between I/O lines, communications lines, and control signals:

- Route the power supply, input and output wiring and communications cables in separate conduits, whenever possible.
- Segregate I/O wiring by signal type, i.e., dry contacts, thermistors, 4–20 ma, etc.
- Allow at least two inches between the monitoring system and I/O wiring ducts.
- Keep communications cable at least five feet (1.5 m) from any electric motors, transformers, rectifiers, generators, arc welders, induction furnaces or sources of microwave radiation.

- Keep communications cable at least 6 inches (15 cm) from AC power lines carrying less than 20 A, at least 1 foot (30 cm) from lines greater than 20 A, and 2 feet (60 cm) from lines greater than 100 KVA.
- If cable is run in a metallic wireway or conduit, keep the communications cable at least 3 inches (8 cm) from AC power lines carrying less than 20A, at least 6 inches (15 cm) from lines greater than 20 A, and 1 foot (30 cm) from lines greater than 100 KVA.

CHAPTER 14

BUILDING ENVIRONMENTAL CONTROL

14.0 INTRODUCTION

This chapter provides a short treatment of the basics of heat and mass transfer. The intent is that the reader will be able to calculate the projected heating costs and establish adequate ventilation requirements associated with maintaining some specific thermal environment for their RAS, since most RAS will be housed within a building. Many people instantly assume that since a RAS is inside a building that the heating costs will be large. The assumption is that if water exchanges are high, then of course the water heating costs will also be high. These assumptions are generally overly pessimistic. In a RAS, water exchange can be managed to be less than 20% per day of system volume and often less than 10%. Under these conditions, water-heating costs will be a small percentage of the total costs of production. Of course, water heating losses will be proportional to water exchange rate. If you had water heating costs with a 10% per day exchange rate of \$0.10/kg, then a 1% exchange rate per pass for a system with a hydraulic retention time (HRT) of 30 minutes would mean a system exchange per day of 48% per day and a heating cost of \$0.48/kg.

Many RAS designers make the mistake of ignoring the ventilation requirements associated with a building that houses an RAS. Ventilation requirements for controlling environmental conditions of humidity, temperature, and CO_2 should be approached in the same way that a ventilation balance is done for chickens or cows to maintain an acceptable air quality environment. For an RAS, we have fish instead of these warm blooded animals. In an RAS, the controlling environmental conditions are typically humidity and then CO_2. Just as we need to do a mass balance for controlling water parameters as described in Chapter 4, a mass balance must be performed for the air space in a RAS with the P terms being moisture, heat, and CO_2. Generally speaking, other air quality parameters can be ignored.

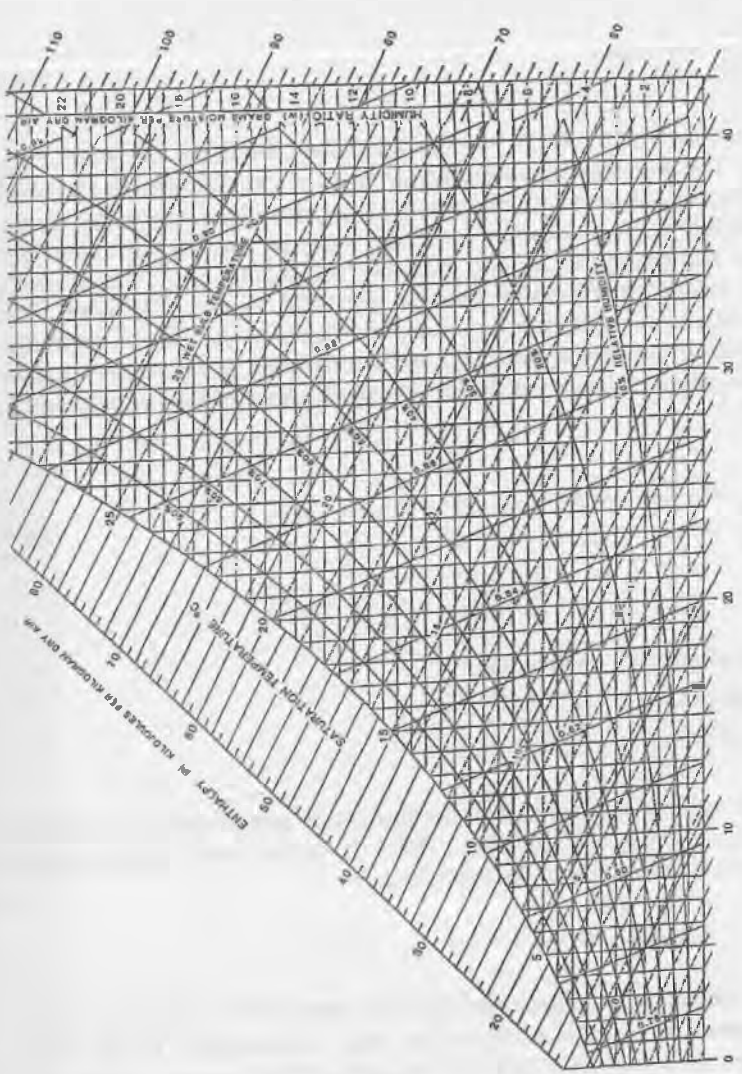


Figure 14.1 Psychrometric chart.

14.1 HEAT TRANSFER

There are two forms of heat transfer:

- Conductive
- Convective

Conductive heat transfer is defined as:

$$Q = \frac{A}{R} \Delta T \qquad (14.1^{a})$$

Convective heat transfer is defined as:

$$Q = \dot{m} c_p \Delta T \qquad (14.2)$$

The value for c_p is 0.24 BTU/(lb°F) or 1.0035 kJ/(kg dry air °C). Generally we think of air in terms of volume of air. The volume of air per unit mass is called the specific weight and is approximately 13.5 ft^3/lb (0.84 m^3/kg) dry air. All the important thermodynamic properties of air are shown in the psychrometric chart given in Fig. 14.1. This figure also shows the grains of moisture per lb of dry air as a function of temperature and relative humidity. (Note there are 7,000 grains in a pound.)

Convection heat loss/gain is proportional to the $\dot{m}$ term in Eq. 14.2 and represents the mass of airflow that is being exchanged or ventilated for the building. For most buildings, air exchange rates caused by wind and natural infiltration will be a minimum of 1 to 2 building volumes of air per hour. As a rule of thumb, maintaining inside relative humidity below 80% will require at least an air exchange of 2 air volumes per hour. Therefore, natural infiltration caused by wind may provide the minimum ventilation requirements for the facility. The point to remember is that you cannot eliminate the heat losses caused by ventilation, since these heat losses will occur as a result of either unplanned infiltration or from proactively controlled minimum mechanical ventilation rates. As a further data point, the required ventilation rates for CO_2 control will also be comparable. Where problems sometimes occur is in extremely tight buildings such as concrete block construction with no windows and only two tight passage doors. Infiltration here will be perhaps as low as 0.5 or less air volumes

[a] Variables defined in List of Symbols at the end of the chapter.

per hour. Under this scenario, CO_2 levels could reach dangerous levels if mechanical ventilation is not used to maintain some minimum ventilation threshold. Heat loss from water flow is calculated the same as shown in Eq. 14.2, except the c_p value for water is 1.0 BTU/(lb °F) or 4.18 kJ/(kg °C).

In insulated buildings, the heat loss through the floor is less than 10% of total building heat loss. This loss term can be estimated based upon the perimeter of the building and whether or not perimeter or floor insulation is used as follows:

$$\frac{Q}{time} = F\,P\,\Delta T \qquad (14.3)$$

The F-term is based upon empirical data and is assigned as:

IP

For Q in BTU, time in hour, P in feet, ΔF in °F

F = 0.81 (4.53 for SI) for floors with no perimeter insulation, BTU/(°F·ft·h)

= 0.55 (3.08 for SI) for floors with 1 inch of rigid insulation, BTU/(°F·ft·h)

SI

For Q in kJ, time in seconds, P in meter, and ΔT in °K

F = 1.38 for floors with no perimeter insulation, W/K-m

= 0.93 for floors with 2.5 cm of rigid insulation, W/K-m

During startup operations, the first winter season will typically cause much higher heat losses than subsequent winter seasons. The first year the soil acts as a large thermal sink and once full, basically eliminates subsequent heat losses or gains. This is why the perimeter insulation is important to prevent heat seepage losses from the thermal envelope.

Yearly Heat Losses or Gains

Determining yearly heating costs or cooling costs requires that an entire year of simulation be used. Heating degree-days (HDD) are used to estimate the amount of energy required for space heating during the cooler seasons. For example, given the value of HDD, the yearly heat loss can be calculated as follows:

$$\frac{Q}{year} = \frac{A}{R} HDD \qquad (14.4)$$

To calculate the HDD's you must first find the mean temperature for the day. The average daily temperature can be estimated as the average of the day's high and low temperatures or calculated according to Eq. 14.5. Equation 14.5 requires monthly average temperature data (the monthly maximum, U_{max}, and monthly minimum, U_{min}, for monthly average temperature); some data is provided in the Appendix. The generic equation to predict any weather variable, U_{day}, for a particular Julian date of the year is based upon the maximum and minimum values of the monthly average values for the particular variable of interest as follows:

$$U_{day} = \left[\left(\frac{U_{max} - U_{min}}{2}\right)(1+\phi)\right] + U_{min} \qquad (14.5)$$

where:

$\phi = -\sin(\lambda - date)$; if $(date - \lambda) < 0$

$\phi = +\sin(date - \lambda)$; if $(date - \lambda) \geq 0$

date=Julian day of calendar year

The λ value is chosen so that the maximum and minimums occur on the appropriate day of the year. These λ values solar radiation and temperature in the northern hemisphere are:

- $\lambda_{solar} = 83$
- $\lambda_{temperature} = 100$

If the mean daily temperature is at or above 65°F (18°C), then the HDD amount is zero (if 65°F is the base temperature). If the mean temperature is below 65°F (18°C), then the HDD amount equals 65° (18°C) minus the mean temperature. For example, if the mean outside temperature was 55°F (13°C) then the HDD amount equals 10 degree-days (5 degree-days in Celsius). Yearly heat losses can be calculated based upon the yearly HDD's for the location of interest.

In equation form:

$$\text{if } T_a < T_{base}$$
$$HDD = T_{base} - T_a \qquad (14.6)$$
$$\text{if } T_a > T_{base} \text{ then } HDD = 0$$

Cooling degree days (CDD) are used to estimate the amount of air conditioning usage during the summer season. CDD's are calculated similarly to HDD's except that you are calculating the degree-days above some base inside air temperature. Think of cooling degree-days as the flip side to heating degree-days.

For easy reference, the fuel contents of various forms of energy are provided in Table 14.1.

Table 14.1 Approximate Fuel Heat Contents

Fuel Type	Heat Content		Units
Natural Gas	1,000	(37,350)	BTU/ft^3 (kJ/m^3)
LP Gas	93,000	(25,800)	BTU/gallon (kJ/Liter)
Fuel Oil	138,000	(38,460)	BTU/gallon (kJ/Liter)
Electricity	3,413	(3,600)	BTU/kWh (kJ/kWh)

14.2 Air Quality Control

Just as we do a mass balance on the fish tanks for pollution loading and the required flow rates to maintain water quality at some minimum target values, we also take the same approach on the thermal air space to control humidity (moisture), temperature, and carbon dioxide (CO_2). For example, a general sensible heat balance on an enclosed space would be depicted in Fig. 14.2

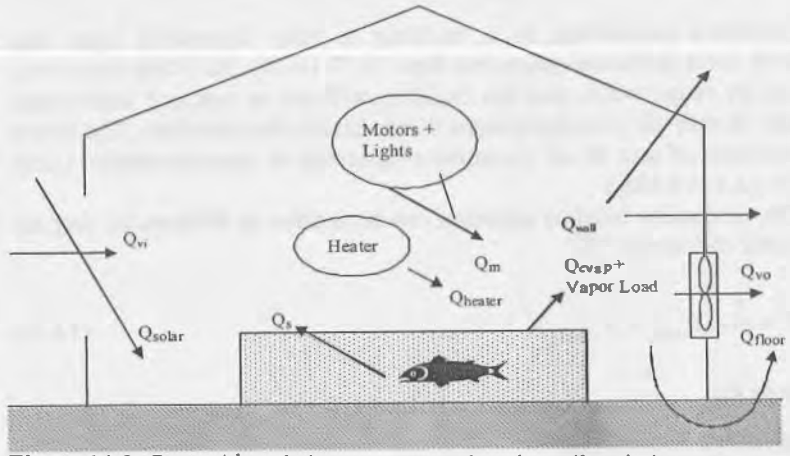


Figure 14.2 General heat balance on an enclosed ventilated air space.

For steady state conditions, the gains of heat must balance the heat losses, or in equation form:

$$Q_s + Q_{solar} + Q_{heater} + Q_m + Q_{vi} = Q_{evap} + Q_{wall} + Q_{floor} + Q_{vo} \qquad (14.7)$$

Heat produced by fish, Q_s, is approximately 1 BTU/hr per lb (2.2 kJ/hr per kg) of body mass. Evaporation rates from free water surfaces are approximately 0.25 inch/day (6 mm).

Evaporation rate, (E, inches per day), can be predicted as a function of wind speed (S, mph), inside relative humidity (RH, %), and vapor pressures of the water, $e_{s,water}$, and the inside air, $e_{d,air}$ (inches of mercury):

(Eqs. 14.8–14.10 valid for IP units only)

$$E = C_{wind}(e_{s,water} - e_{d,air}) \qquad (14.8)$$

$$C_{wind} = 0.44 + 0.118S \qquad (14.9)$$

$$e_{d,air} = RH \cdot e_{s,air} \qquad (14.10)$$

From Chapter 3, we know the CO_2 production rates are proportional to oxygen consumption rate (roughly 1.4 times the oxygen consumed).

The moisture production in a building is very dependent upon the humidity level in the air space (see Eqs. 14.7–14.10). At 100% humidity, there is no evaporation, and the building will rot or rust and deteriorate quickly, as will all your equipment that is inside the structure. The latent heat content of one lb of moisture evaporated is approximately 1,050 BTU/lb (2,440 kJ/kg).

The air quality balance equation can be written as follows for any air parameter of interest "X":

$$P = \dot{m}\,(X_{inside} - X_{outside}) \tag{14.11}$$

EXAMPLES

CONDUCTIVE HEAT LOSS

Wall area is 100 ft², R_{value} = 10°F ft²/BTU, inside temperature is 80°F and outside temperature is 50°F. Calculate hourly heat loss:

$$Q\left(\frac{BTU}{hr}\right) = \frac{A}{R}\Delta T = \frac{100\,ft^2}{10\dfrac{hr\cdot{}^\circ F ft^2}{BTU}}(80-50)^\circ F = 300\frac{BTU}{hr}$$

Metric SI

$$Q(W) = \frac{A}{R}\Delta T = \frac{9.29\,m^2}{1.761\dfrac{m^2\cdot{}^\circ C}{W}}(26.7-10.0)^\circ C = 88W$$

CONVECTIVE HEAT LOSS-WATER

Calculate the heat loss from exchanging 1 gal/min (3.785 L per minute) when the water used to replace the water being discharged (makeup water) is 45°F (7.2°C) (generally this is well water or deep groundwater temperatures) and the inside tank water temperature is 75°F (23.9°C).

$$Q = \dot{m}c_p\Delta T = 1\frac{gal}{min}\cdot 8.34\frac{lb}{gal}\cdot 1.0\frac{BTU}{lb\cdot{}^\circ F}\cdot(75-45)^\circ F\cdot 60\frac{min}{hr} = 15{,}012\frac{BTU}{hr}$$

Metric SI

$$Q = \dot{m}c_p\Delta T = 3.785\frac{L}{min}\cdot 4.187\frac{kJ}{kg^\circ C}\cdot(23.9-7.2)^\circ C\cdot 60\frac{min}{hr} = 15{,}880\frac{kJ}{hr}$$

CONVECTIVE HEAT LOSS-AIR

Calculate the heat loss per day due to air ventilation from a building that is 100 x 100 x 10 ft (length, width, and ceiling height) when the outside air temperature is 50°F and the inside air temperature is 80°F and the ventilation rate is 3 air volumes per hour.

$$Q = \dot{m}c_p\Delta T = 3\frac{vol}{hr}\cdot 100\cdot 100\cdot 10\frac{ft^3}{vol}\cdot\frac{lb_{air}}{13.5\,ft^3}\cdot 0.24\frac{BTU}{lb\cdot{}^\circ F}\cdot(80-50)^\circ F\cdot 24\frac{hr}{day}$$

$$= 3{,}840{,}000\frac{BTU}{day}$$

Metric SI

$$Q = \dot{m}c_p\Delta T = 3\frac{vol}{hr}\cdot 2{,}825m^3\cdot 1.19\frac{kgair}{m^3}\cdot$$

$$1.007\frac{kJ}{kg^\circ C}\cdot(\Delta T = 16.67)^\circ C\cdot 24\frac{hr}{day} = 4{,}063{,}000\frac{kJ}{day}$$

COST TO HEAT AIR

Calculate the heating cost associated with the above air heat loss example if the building is heated with LP gas that has a heat content of 92,000 BTU/gal, the burn efficiency is 85%, and the cost of the LPG is $0.80/gal.

$$\$/day = 3{,}840{,}000\frac{BTU}{day}\cdot\frac{gal_{LPG}}{92{,}000BTU}\cdot\frac{\$0.80}{gal_{LPG}}\cdot\frac{1}{0.85} = \frac{\$39.28}{day}$$

Metric SI

$$\frac{\$}{day} = 4{,}051{,}200\frac{kJ}{day}\cdot\frac{L_{LPG}}{25{,}643kJ}\cdot\frac{\$0.21}{L_{LPG}}\cdot\frac{1}{0.85} = \frac{\$39.03}{day}$$

VENTILATION RATE FOR MOISTURE CONTROL

Calculate the required ventilation rate to maintain an inside relative humidity of 80% at an air temperature is 68°F, if the outside air temperature is 41°F and 70% relative humidity. You have 1,000 ft^2 (93 m^2) of free water surface exposed (assume 0.25 inches, 6 mm, of water is evaporated per day from a free water surface).

$$P_{water}(lb/day) = 1{,}000\,ft^2_{water} \cdot \frac{0.25inch_{water}}{day} \cdot \frac{1\,ft}{12inch} \cdot \frac{62.4lb_{water}}{ft^3_{water}} = 1{,}300\frac{lb_{water}}{day}$$

$$W_{inside}(68°F\ \&\ 70\%) = 0.0102\,{}^{lb_{water}}\!/_{lb_{air}}$$

$$W_{outside}(41°F\ \&70\%) = 0.0036\,{}^{lb_{water}}\!/_{lb_{air}}$$

$$\dot{m} = \frac{P_{water}(lb/day)}{(W_{inside} - W_{outside})} = \frac{1{,}300\frac{lb_{water}}{day}}{(0.0102-0.0036)\,{}^{lb_{water}}\!/_{lb_{air}}}$$

$$= 196{,}970\frac{lb_{air}}{day} \cdot \frac{13.5ft^3}{lb_{air}} \cdot \frac{day}{1{,}440\min} = 1{,}846\,cfm$$

Metric SI

$$P_{water}(kg/day) = 93m^2_{water} \cdot \frac{6.35mm_{water}}{day} \cdot \frac{m}{1{,}000mm} \cdot \frac{1{,}000kg_{water}}{m^3} = 590\frac{kg_{water}}{day}$$

$$W_{inside}(20°C\ \&\ 70\%RH) = 0.0102\frac{kg_{water}}{kg_{air}}$$

$$W_{outside}(5°C\ \&\ 70\%RH) = 0.0036\frac{kg_{water}}{kg_{air}}$$

$$\dot{m} = \frac{P_{water}(kg/day)}{W_{inside} - W_{outside}} = \frac{590\frac{kg_{water}}{day}}{(0.0102-0.0036)\frac{kg_{water}}{kg_{air}}}$$

$$= 89{,}394\frac{kg_{air}}{day} \cdot \frac{0.84m^3}{kg_{air}} \cdot \frac{day}{1440\min} = 52\frac{m^3}{\min}$$

14.3 BUILDING CONSIDERATIONS

MATERIALS

In an aquaculture setting, your buildings inside air conditions will often be a warm moist air environment. Under such conditions of high humidity, a drop in inside temperature (referred to as dry bulb temperature or T_{db}) or when warm moist air comes in contact with a cooler surface, the air will drop in dry bulb temperature. If the resulting air temperature is below the dewpoint temperature (T_{dp}), then condensation will occur. Figure 14.1 provides a psychrometric chart that can be used to determine when condensation would occur for specific inside T_{db} and relative humidity (RH) conditions. For example, if the design inside T_{db} were 80°F (26.7°C) and 75% RH, then the T_{dp} is 71.5°F (21.9°C) at 100% RH. The amount of moisture (sometimes called humidity ratio, W) in the air remains the same between these two conditions (W = 0.0168 lbs moisture per lb dry air or .00762 kg/kg dry air).

Using adequate wall and ceiling insulation can prevent condensation. To accomplish this, you would have to work from an expected outside design temperature and then install enough insulation in the wall to prevent the surface from dropping below 71.5°F (21.9°C) in the previous example. Consult just about any heat transfer text for more information in this regard.

For quick reference, the formula to calculate minimum R-value for a wall to prevent condensation is:

$$R_W \geq R_{inside}\frac{t_{db\,inside} - t_{db,outside}}{t_{db,inside} - t_{dp}} \tag{14.12}$$

where R_{inside}

- 0.12 m^2°C/W Vertical wall (high surface emittance)
- 0.11 Horizontal surface heat flow upward
- 0.16 Horizontal surface heat flow downward

For low emittance walls such as foil faced, the R_{inside} will be about twice the above values.

For non-free convection coefficients pertaining to wall surface resistance such as outside walls or inside walls in forced convection conditions, the R_{inside} will be 0.030 to 0.04 m^2°C/W. (Note the conversion from SI to PI is: 5.678 hr ft^2 °F/BTU per m^2°C/W).

We strongly recommend against using any type of fibrous insulating materials, e.g., fiberglass batting, paper based materials, rock wool.

These materials lose 90% of their insulating ability after absorbing only 10% of their initial weight in moisture. We recommend using rigid board insulation, e.g., polystyrene blue board. Ideally, the board material should be covered by a metal surface, but where this is not done, use foil faced board-insulating materials. The Appendix provides Table A-14 of common building materials and R-values.

> **"Rule of Thumb"**
>
> Avoid fibrous insulating materials. Use rigid board insulation.

MOISTURE CONTROL

Consideration should be given to preventing moisture migration into walls and for adequate ventilation in attic spaces. Moisture transfer can be calculated as follows:

$$w = \frac{(P_{wv,inside} - P_{wv,outside})}{R_{H,0}} \tag{14.13}$$

The analogy to Eq. 14.13 is steady-state heat transfer by conduction. Vapor pressure difference is analogous to temperature difference, permeance is analogous to thermal conductance, and permeability is analogous to thermal conductivity. The concepts of simple parallel and series resistance circuits also apply to water vapor diffusion as they do to heat diffusion and transfer.

It is generally impossible to prevent moisture migration. Most migration is not through the materials themselves, but from air leakage into the attic space from the warmer area below where animals and people are. During construction and after, pay particular attention to closing all cracks and crevices, particularly those created by installing fixtures into the ceiling. Flush mounting of electrical fixtures is preferred to recess mounting, since no cracks or break in the ceiling materials are created when doing flush mounts.

For attic spaces, 1.0 square unit ventilation area for each 300 square units of ceiling area should be provided. Since louvers will have rain shields and screening, provide 2.25 times the area required or 2.25 square units for each 300 square units of ceiling area.

Table 14.2 provides some permeability values for common building materials.

Table 14.2 Permeability for Various Building Materials

Material	Perm-inches*	Perms**
Air	120	
Gypsum board		50
Interior plywood, 1/4 inch		1.9
Exterior plywood, 1/4 inch		0.7
Pine wood	0.4–5.4	
Concrete	3.2	
Roll roofing		0.05
Aluminum paint		0.3–0.5
Latex paint		5.5
Mineral Wool	116	
Blanket insulation and asphalt paper		0.04
Expanded Polystyrene		
Extruded	1.2	
Bead	2.0–5.8	
Polyurethane		
Polyethylene 4 mil		0.08
Polyethylene 8 mil		0.04

*To obtain (gram-m)/(24 hr m^2 mm Hg) multiply by 0.017
** To obtain (grams)/(24 hr m^2 mm Hg) multiply by 0.66

> **"Rule of Thumb"**
>
> - Use 1.0 ft^2 of net louver area for each 300 ft^2 of ceiling (1 m^2/300 m^2).
> - Increase louver area by a factor of 2.25 to account for screening and louver blockage.

LIST OF SYMBOLS

A	Area, ft^2 (m^2)
c_p	Specific heat of air, BTU/(lb°F) (kJ/kg°C)
date	Julian day of the year as a number
F	Perimeter heat loss term, BTU/°F·ft·h (W/K·m)
$\dot{m}$	Airflow rate, lb/hr (kg/hr)
P	Perimeter length of exposed walls, ft (m)
$p_{wv,inside}$	Vapor pressure inside air, inches mercury water gauge (mmHg)

$p_{wv,outside}$ Vapor pressure outside air, inches mercury water gauge (mm Hg)
Q Heat loss or gain, BTU (kJ)
Q_s Sensible heat production of fish, BTU/h (kJ/h)
Q_{solar} Solar heat gain, BTU/h (kJ/h)
Q_{heater} Sensible heat added by space heaters, BTU/h (kJ/h)
Q_m Sensible heat added by motors and lights, BTU/h (kJ/h)
Q_{vi} Sensible heat ventilated into air space, BTU/h (kJ/h)
Q_{evap} Rate of sensible heat converted to latent heat via evaporation, BTU/h (kJ/h)
Q_{wall} Sensible heat conducted from the space through walls and ceiling, BTU/h (kJ/h)
Q_{floor} Sensible heat lost through the floor, BTU/h (kJ/s)
Q_{vo} Sensible heat ventilated out of air space, BTU/h (kJ/h)
R Thermal resistance, (hr°F ft^2)/BTU (m^2·°K/W) (see the Appendix A-14 for a listing of R-values for common materials, from ASHRAE 1985).
R_{H2O} Resistance to water vapor flow; inverse of permeance or perms, hr ft^2 inch Hg/grains (g-m/(24 h m^2 mm Hg))
RH Relative humidity, %
T Temperature, °F (°C)
T_{base} Temperature base, usually 65°F (18.3°C)
T_a Average temperature, °F (°C)
T_{db} Dry bulb temperature, °F (°C)
T_{dp} Dewpoint temperature, °F (°C)
U_{max} and U_{min} Maximum and minimum monthly average values for particular weather variable
W Rate of moisture movement, grains/hr (grams/hr)
X_{inside} Inside air quality parameter, lbs per lb dry air (kg/kg)
$X_{outside}$ Outside air quality parameter, lbs per lb dry air (kg/kg)
ΔT Temperature difference inside air film to outside air film, °F (°C)
λ_{solar} Weather model constant for solar radiation (83)
$\lambda_{temperature}$ Temperature weather model constant for dry bulb temperature (100)
ϕ Argument used in weather model based upon Julian date

CHAPTER 15

SYSTEM MANAGEMENT AND OPERATIONS[1]

15.0 INTRODUCTION AND SITE SELECTION

Before you start a fish farm, you need to select a location. Selecting a location will also define a political district where you will have to meet all the laws and regulations covering your intended fish farming operation. So, step one is to determine if you can meet the laws and if you can, whether this means you still want to locate your farm at this location. Make sure you understand all the rules. The next step is to identify if you have sufficient water and other logistical support structures, e.g., roads, bridges with adequate loading capacity, utilities and the rates associated with each. We provide a checklist of factors that you should investigate prior to purchasing a site (see Appendix section "Factors to Investigate Prior to Site Selection")

During almost all discussions of intensive recirculation aquaculture systems, the focus is on the culture tanks, filtration systems, aeration/oxygenation systems, and the species being cultured. Yet the system's support components, which are just as important and in some cases critical to commercial success, are rarely mentioned. Support components include all of the other parts of a recirculating system that are necessary for its profitable operation. How well these support systems are designed, integrated into the operation, and managed often determines whether a recirculating system survives commercially or not. Many of these supporting systems are common to all fish production facilities; some requirements are unique to recirculating systems. One common characteristic is that they are often ignored in estimating culture system construction costs. As a result, support systems are often installed late in the construction phase or as an afterthought subsequent to a disaster. When this happens, funding is limited and typically the least expensive support system is installed, regardless of its reliability or

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whether it is appropriate to the system. Such false economy usually compounds problems in an already marginal operation. The result of such activities is then just a matter of when, and not if, the system will fail.

The list of support components necessary for an intensive recirculating aquaculture system (RAS) is a reflection of the level of sophistication, the interplay of upfront capital investment versus daily operational costs, size of operation, number of employees, geographic location, and numerous other parameters. A short list includes such items as:

- Backup Systems
- Laboratory Facility
- Quarantine Area
- Waste Management
- Storage – Feed, Chemicals, Product
- Handling of fish, both live and post slaughter
- Labor
- Access

The objective of this chapter is to identify some of the needed support systems with no attempt to be all inclusive or to prioritize. Each recirculating system will have it own special needs and the priority of needs for one system is often different from another system. Thus, the system design engineer and the manager must include the required support systems pertinent for any specific production unit.

15.1 BACKUP SYSTEMS

In science, a hypothesis only becomes a law after being tested and observed countless times. Murphy's Law is an excellent example of this concept. It states simply: If anything can go wrong, IT **WILL!** This is the entire design rational behind backup systems and even backup systems for backup systems. During design, construction, and operation, it is critical to try to imagine the worst-case-scenario! Because if it can happen (and often when you think it cannot possibly happen), IT WILL! Anticipate! Plan! Train! Respond!

"Rule of Thumb"
Backup Systems
Anticipate! Plan!
Train! Respond!

One of the primary systems requiring backup is the electrical power supply, which is required to operate pumps, aeration systems, instrumentation systems, and to perform a variety of other functions in a recirculating system. Failure of the electric supply can have devastating effects in a matter of minutes, especially in heavily loaded systems. Backup electricity can be provided by a generator that is fueled with gasoline, diesel, or natural or propane gas. Backup generators are a critical must for any commercial (for-profit) system. Several commercial manufacturers can supply turnkey systems over a wide range of power requirements, Fig. 15.1.

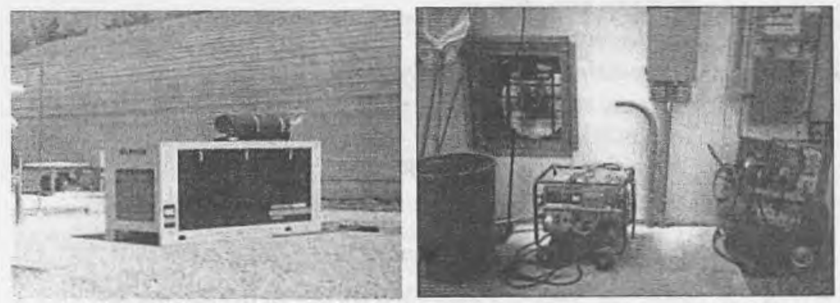


Figure 15.1 Backup generators: dedicated diesel unit and a portable standby unit.

The cost of the generator is directly related to the generator size, i.e., the power in kilowatts that is generated. The generator size required for backup power is determined by the loads that are critical to the maintenance of good water quality or otherwise support the survival of the fish in the culture systems during power blackouts. Typically, this includes powering such items as the circulating pumps, aerators or blowers, the data acquisition system and building emergency lightning. Usually, these loads are handled by a separate emergency circuit breaker panel. The capability of the backup generator in terms of voltage (120–240 V AC), frequency, current, and phase (single or three phase) will be determined by these essential support requirements.

One of the important design parameters in specifying a backup generator is whether it will or will not have automatic starting capabilities. Automatic starting systems monitor incoming power lines and when the external power supply goes down, the generator automatically starts, so power is always available to critical components. Automatic systems or cut-over switches are expensive, but are necessary when personnel are unavailable during certain periods of the day or are unable to get to the facility rapidly enough to carry out manual emergency measures. An example autotransfer switch with the emergency circuit breaker panel on the right is shown in Fig. 15.2.

An automatic transfer switch will cost roughly 35% of the total cost of the backup generator system, but is generally well worth the investment if continuous manual coverage is not possible. If you use an auto-transfer mechanism, make sure that protocols are in place to protect the fish if the auto-transfer fails. Sometimes, the alarm sensors will "sense" that power has been restored when in fact it has not. Three-phase systems are particularly problematic and tend to fail more often in the transfer operation than single phase systems. Remember, an auto-transfer is not nearly as reliable as 24-7 coverage by humans and a manual transfer switch.

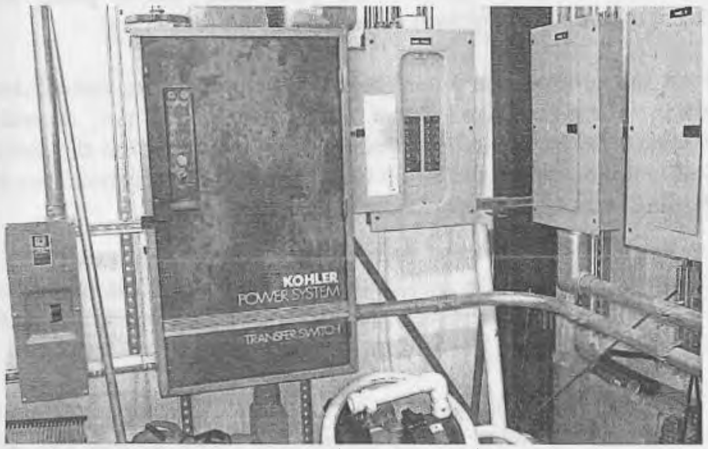


Figure 15.2 Generator automatic transfer switch and emergency circuit breaker panel.

Even people are not 100% reliable as a backup system. They must be properly trained to respond to emergency and to monitor continuously. It is a good practice for a night shift person to have duties that require them

to remain active throughout their shift. Intentionally schedule activities that keep them busy, e.g., water chemistry measurements, and some hand feeding.

Regardless of the generator system chosen, an automatic or manual transfer switch will be required that disconnects the power company supply lines and connects the generator lines. These switches are required to prevent feedback of power into the power company grid from the system generator. Feedback into the power company grid is a safety hazard to power line workers repairing lines. In addition, if feedback occurs, shorts or loads on the power grid can overload the generator and burn it out. Whether manual or automatic, transfer switches are expensive and can cost as much as the basic generator. It is important to work with the local power supplier to determine exactly what their requirements are and in some cases, whether equipment can be leased from them.

"Rule of Thumb"

There is NOTHING more reliable than **"24-7"** coverage by a person who can flip a manual transfer switch to backup power when you lose power!

Generator maintenance is an absolute must! If power outages are infrequent, the backup generator may sit for long periods of time between uses. Fossil fuel engines, whether diesel or gasoline, not started for prolonged periods tend to be difficult to start. Failure of the backup generator's engine to start due to dead batteries, low fuel levels, or other reasons will lead to catastrophic fish loss and perhaps the end of the business itself! Most commercial models are designed to be operated for a specified period every few days. Smaller generators should be periodically started and maintained in top running condition at all times.

Another style of backup generator is shown in Fig. 15.3. Note in the figure that the generator set has a roof over it (not shown) and that the generator set is well removed from the nearest wall. This provides fire protection, prevents heat buildup around the generator set, and provides a measure of sound reduction.

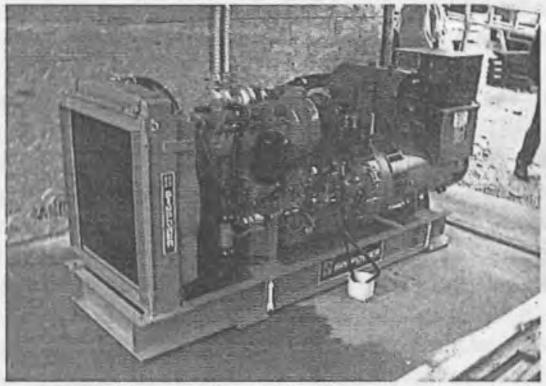


Figure 15.3 Backup generator.

Sizing a Backup Generator

It is first useful to have a basic understanding of the terms used to calculate loads and describe unit features. Here is the minimum information that you should understand:

- Standby Power is power available at variable load capacities for the duration of the main power network interruption.
- Prime versus stand-by power. According to ISO 3046 ratings, baseload power is defined as power available for continuous operation, with 10% overload available for 1 hour in every 12 hours of operation. Backup generators can be designated as a prime power unit (this is your primary source of power at this point) if the generator is selected for peak load operation at variable loads for an unlimited number of hours in place of the main power network.
- Transfer switches are the devices that switch the electrical feed from line power to the generator set. They can be automatic or manual.
- kVA is 1,000 volt-amps and is a power rating method used to describe the size of the generator. This number is used to size the generator and not the horsepower of the motor on the gen-set. The equations to determine the kVA of the generator are:

$$kVA_{1-phase} = \frac{Volts \cdot Amps \cdot PF}{1{,}000} \qquad (15.1)$$

$$kVA_{3-phase} = \frac{Volts \cdot Amps \cdot PF \cdot 1.73}{1{,}000} \qquad (15.2)$$

where PF is the power factor for the motor

Values for PF range from 0 to 1.0; typically use 1.0. PF accounts for the reduction in power output due to inductive reactance. When the device is all resistance, such as in an electric water heater, the PF is 1.0. When the electrical circuit contains a coil, which produces inductive reactance, the PF will be less than 1.0, but will be typically above 0.8. For exact information, you must contact the motor manufacturer.

Testing the Back-up Generator

Your back-up generator must work when the critical time comes. You will not have time to refine your protocols once you have lost power. The acid test is to go over to your main power panel and disconnect the incoming line power. Then check the following:

- Is all the critical equipment running (or was there insufficient power to start all the critical units at once). If not, consider installing delay timers on specific equipment so that the entire load is not activated at startup (startup amp requirements may be two to five times running load amps).
- Let the system run under full load for 6 hours at least twice a year to be sure that the generator set is adequate. This will allow any problems to be identified and corrected before a crisis occurs.

Final advice on this subject: make sure your backup generator set is installed and has been tested prior to your fish being placed into the facility.

Loss of Oxygen

More fish probably die in recirculation systems due to a lack of oxygen than to any other single cause. Backup oxygen systems are a basic requirement for a culture system to be economically feasible. Because oxygen availability is so critical to heavily loaded culture

systems, often a three tier emergency oxygen supply is prudent. There are many types of oxygen supply systems used in RAS and the type of emergency backup system needed varies with the primary oxygen supply system used. One of the simplest and most cost-effective emergency oxygen systems uses an oxygen tank, either liquid or gas, connected to micro-diffusers in the culture tank through a normally open, electrically operated solenoid valve (see Fig. 15.4). When electricity is applied to the solenoid valve, the valve is closed and oxygen is provided by the regular oxygen supply system. When the power goes out for some reason or the system is manually operated, the solenoid opens, allowing oxygen from the tank to flow into the culture tank. It is important for any backup system, but especially for oxygen systems, that they are automatic and engage at the first sign of a potential problem.

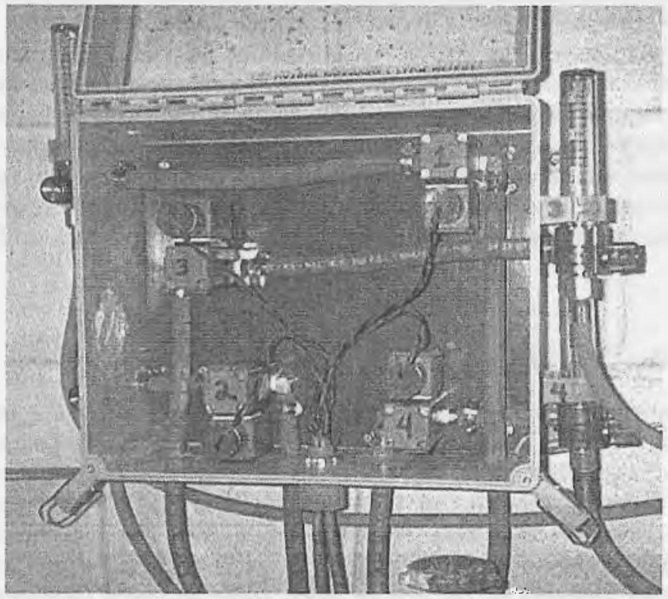


Figure 15.4 Solenoid oxygen backup system with oxygen flow meters for monitoring.

Have your oxygen back up systems in place. These units, whatever they are, need to be in or near the tank so that on a moments notice, you can activate them. Once you lose power (flow) you have only minutes to respond to protect all the tanks in the facility.

"Rule of Thumb"

You have 5 to 15 minutes to restore oxygen to all of the tanks in the facility, once you've lost power or water flow. Can you do it?

So, do a dry run before the crisis occurs. Can you put all fish tanks on their backup oxygen systems in 5 minutes? If not, re-evaluate and re-plan until you can.

15.2 Laboratory Facilities

The size and sophistication of the laboratory space needed for an aquaculture facility will vary with the size and complexity of the recirculation system. However, every recirculating aquaculture production system will require at least a minimum amount of space set aside for laboratory analysis. At a minimum the laboratory will include equipment for water quality analysis, a microscope for fish health management, a refrigerator for chemicals and samples, and a computer for data analysis and storage.

Water quality monitoring and control is a routine task in any aquaculture facility. The amount of laboratory space and equipment needed for this work will vary with the methods used, facility size, and the frequency of sampling. Smaller fish farms can often get by with commercial water quality test kits that are available from several manufacturers. They use pre-packaged chemicals, indicator strips, or color comparison techniques. They are convenient, relatively inexpensive per analysis, and provide sufficient accuracy for small production facilities with low stocking densities. More importantly, they require very little laboratory space.

As the number of samples and the corresponding risks to production increase, the sophistication of the analysis also needs to increase. Several important water quality parameters can be monitored using electrode sensors that provide an electrical output signal proportional to the water quality parameter being measured, such as temperature, pH, dissolved

oxygen, conductivity, and oxygen reduction potential (ORP). Portable meters do require that someone physically go to each sampling point and manually measure and record the required water quality parameter at whatever is the desired frequency. Labor costs for manual monitoring can be high for large system, but economical for small systems, in comparison to the equipment costs for dedicated systems.

In heavily stocked systems, dedicated systems can be used to both monitor and control critical water quality parameters, such as temperature, pH, and dissolved oxygen. Commercial systems are available that will monitor selected parameters, and also provide an alarm when a parameter deviates from the preset minimum and maximum. Combined with the necessary computer hardware and software, real time water quality data becomes available to the system's manager. This provides accurate information for management and historical data for system performance evaluation and analysis.

As the sampling protocols become more sophisticated, a higher quality laboratory will be needed, Fig. 15.5. Currently, for example, there are several commercially available laboratory spectrophotometers designed for water quality analysis. These dedicated pieces of equipment are able to analyze for a wide range of critical water quality parameters using pre-packed chemicals and simple laboratory procedures. At this point, the laboratory should be equipped with a sink, refrigerator, and computer, and have adequate room for equipment storage and personal work areas.

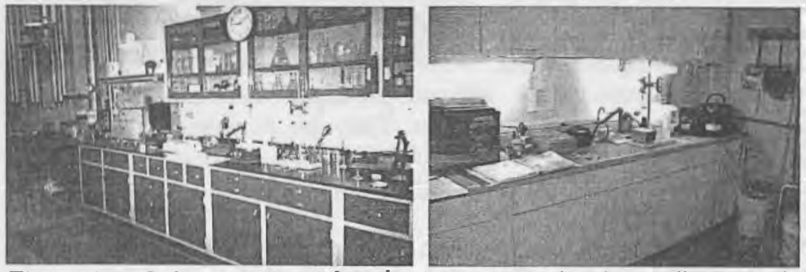


Figure 15.5 Laboratory space for a large commercial and a smaller operation.

15.3 Quarantine Facilities

Many of the disease outbreaks occurring in recirculating systems come from diseases introduced on or in fish purchased from outside the system. Disease introduction can be minimized by quarantining incoming fish for one to a few weeks prior to introducing them into the recirculating system. During this period any disease problems can be treated without contaminating the recirculating production system. See Chapter 16 for more details on quarantining.

Quarantine areas require space and must be located such that the incoming fish are never in close contact with the growing systems. Personnel traffic must never be allowed to move from the quarantine area directly into the area containing the growing systems. Permanent physical barriers must make such movement impossible. Where possible, the quarantine area should be in a separate building from the main production systems. The size of the quarantine area is a function of the management techniques used in the facility. The quarantine area must be large enough to house all of the fish entering during one quarantine period, whether it is one week or several weeks. The water supply must be adequate for the volume of fish in the quarantine area and separate waste discharge should be provided. Further information on quarantine procedures and biosecurity issues are discussed in more detail in Chapter 16.

15.4 Waste Management

Aquaculture waste discharge regulations are currently in a state of flux. However, it is clear that aquaculture producers will be required to meet discharge regulations and these regulations probably will become more stringent in the future (Chapter 6). Thus, waste management facilities and management methodologies will be an important component of any aquaculture facility put on-line in the future. Because recirculating systems do not use as much water as many other types of aquaculture systems, their waste management problems may not be as severe, but there is waste that must be disposed of or put to use in an environmentally beneficial or at least benign way. See Chapter 6 for more details.

Waste disposal needs for an aquaculture facility depend on the system size, species cultured, feed used and other variables. The major problem is removing the wastes from the culture water. There are

mechanical, biological and chemical methods to accomplish this removal, and a variety of specific implementing systems. The best technique to use depends on the waste characteristics, concentrations, and form in which the waste is found. These filtration systems are covered extensively in Chapter 5 Solids Capture. Once the waste is removed from the culture water, it must be disposed of or used for some useful purpose. Solids from fish culture systems contain considerable nitrogen and phosphorous, which are useful nutrient elements in fertilizers. Thus, one disposal technique is to use the solids for fertilizers, by spreading it on agricultural land. Composting for later use as a soil amendment, mulch, or fertilizer can be used for both solid and liquid fish wastes. Holding facilities may be necessary to contain the wastes between periodic disposal cycles. Such facilities must contain the wastes in an acceptable manner and must prevent development of odors and other noxious nuisances.

Nearly all aquaculture facilities will suffer some mortality, although under normal operation the volume of dead fish will be low. Nevertheless, if mass mortalities occur due to oxygen stress, disease, or some other reason, the volume of dead fish that must be disposed of can become high. These fish must be disposed of in an environmentally acceptable manner, and they cannot create noxious odors or pose sanitation or health risks to either humans or other fish. Acceptable disposal techniques will depend on the volume of dead fish, the land use near the facility, the depth of the groundwater table and other factors. In the event of a mass mortality, lack of planning for disposal of dead fish can be embarrassing at best and may result in a legal morass. If fish are processed on-site, processing waste must also be disposed of or used. Some of the options for large quantities of fish waste are composting, anaerobic digestion, burying, or moving to a landfill. The key is to be prepared ahead of time.

15.5 Storage—Feed and Chemicals

There are several obvious materials that must be temporarily stored at any aquaculture production facility including: 1) chemicals for disease treatment and for running water quality tests, 2) finished product, and 3) feed. There are several potential uses for these chemicals in recirculating systems, although only a limited number are available for disease treatment of fish. However, those chemicals that can be used must be stored in an appropriate manner. Some require refrigeration, while others

require only dry storage conditions. All of these chemicals should be secured against accidental use and/or theft.

Chemicals used for water treatment are probably more common than disease treatment chemicals in recirculating systems. Typically, flocculants, disinfectants, and cleaning compounds are used in or around recirculating systems and space must be allocated for their storage. Normally such storage areas must be ventilated, dry, and relatively secure. Chemicals are necessary for calibration, titration, and other applications when measuring water quality. Most of these chemicals can be stored in a dry, secure area. Occasionally, a chemical is needed that is corrosive or volatile; these type of chemicals must be stored in an appropriate safety cabinet or refrigerated safety cabinet.

OSHA material safety data sheets (MSDS) describing the chemical and its effects must be available for all workers. EPA labeling and disposal requirements must be met. CVM (FDA) withdrawal times must be adhered to.

15.6 Fish Product Handling

Whatever the production facility, it will produce some final product, usually either live fish ready for sale, or dead fish in the round or processed to some extent (at least gutted, in most cases). Temporary storage must be available for the product. If live fish are to be sold, a holding tank and/or a depuration may be necessary. Often fish are graded (see below) from the production tanks just prior to sale. In this case, the fish to be sold are often collected and held in a separate holding tank that is readily accessible to the truck to be used for transportation.

Grading

Grading is generally accepted as a way to improve growth rates by eliminating negative interactions between fish of different sizes. Grading also allows for a more accurate feeding regimen, feeding the proper feed particle sizes, and it makes harvesting easier to plan and to carry out. The less variability in fish sizes across the cohort, the less grading that will have to be done. Generally, the less deviation in sizes, the more marketable the fish will be to a growout facility and to the processor,

although some processors may want different sizes or a mix of sizes from time to time, to meet their market needs.

Some Norwegian salmon farmers use grading to cull the smallest, least efficient feeders and might remove as much as 50% of the population. Maintaining inefficient feeders can create a 30–50% increase in production costs; therefore, these fish should be removed as early as possible. Culling decisions will need to be made by the manager, based on experience with the facility's production. If it is not clear how well various size ranges of fish are converting feed, a small percentage of the smallest fish from a single tank can be removed. Feed conversion or other parameters can then be monitored to determine whether the initial cull improved production efficiencies.

There are many fish graders commercially available. Whatever the system used, facilities, and/or equipment are needed for grading. The type of grading equipment needed depends on the system design, species, management methods, and other factors. Grading procedures are discussed in detail later in the chapter.

Grading Method

The box grader is often used to grade fingerlings. It consists of a floating box that contains an adjustable or replaceable set of grader bars. In most cases, the appropriate bar width will be determined by trial and error. After grading a few times, grader size may be correlated with condition factors estimated from weight and length samples, and grading can be planned more accurately. The production notebook should contain records of fish size and grader bar width so that benchmarks relating fish size and weight or condition factor to grader width can be established.

Fish to be graded are placed in the top of the grader while it floats in the tank. The small fish pass through, and the larger fish that are trapped inside are emptied into a separate tank. Gently raising and lowering the grader up and down in the water can facilitate the grading process. Grading of fish into two size groups is most easily done using three tanks, but can be done with two. When three tanks are used, the source tank is gradually emptied as the other two tanks are filled with small or large fish.

When two tanks are used, fish are crowded to one side of the tank using a partition, such as a hinged screen. A portion of the fish are then netted and placed into a grader floating on the other side of the partition.

The small fish fall through the grader into the empty half of the first tank. The grader is then lifted from the water and the large fish remaining in the grader are placed into the second tank. The empty grader is then returned to the first tank and the process continues until all fish are removed from the crowded side of the first tank. Ideally, all grading and sampling for growth estimates should be finished in one or two days so that fish growth during the days of grading does not negatively affect data quality.

> **"Rule of Thumb"**
>
> Important: Grading is stressful. Remember to take fish off feed for 24 hours prior to grading.

15.7 Transporting Live Fish

Fish transport is life support for the animals. In order to have a successful business, you have to keep your fish alive during shipping. When using RAS technology, this frequently means learning how to move healthy fish into the system as well as shipping them to market in live form. It makes little sense to spend time and effort to develop and operate an efficient system if you don't also learn how to move animals without causing them harm. Many times fish being transported live are treated poorly during shipment, which leads to stress, disease, and on-going mortality for several weeks or even months.

The first consideration when live hauling is to know that the species you are moving will affect how you move them. Many species require different water temperature and loading rates for successful transport.

Live shipment is involved in two aspects of the business:

- Fingerling transport from supplier to the culture facility
- Shipment of product to live markets

In the first instance, fingerlings must be transported from the supplier to the grow-out facility. They must arrive in good condition to minimize stress-related damage and mortality. It is important to deal with reputable suppliers who have a track record of producing quality fingerlings. Stressed or diseased fingerlings are no bargain at any price. They can lead to disease organisms entering the culture facility that can cause many problems for the operator and be extremely difficult to suppress.

Health certification should always be provided in accordance with state laws and regulations. This usually requires the shipper, or fingerling supplier, to obtain health certificates and submit copies to the receiving

state authorities prior to shipment. Never allow fish to be brought into your system without the proper permits and health certification. Proper quarantine procedures should be followed upon arrival to the facility (see Chapter 16 Fish Health Management). Secondly, if the fish are marketed live at the end of their growth cycle, the product must be shipped so that the animals arrive at market undamaged and free from stress that can induce disease and premature death. Stress resulting from poor hauling practices can lead to quick mortality, which will result in a poor reputation for you as a supplier.

It is important to minimize stress during handling and transport. Fish respond to perceived threats, such as handling, by releasing adrenaline that is carried by their bloodstream. Then steroids are released that affect glucose levels, heart rates, and red blood cell counts. Digestion may cease for a time as well. There are a host of physiological changes that follow that can affect the animals for over a day and can result in sick fish at the delivery point.

Poor handling practices are frequently noted by infections occurring in the fish within a short time. Research indicates that the initial crowding and netting brings on most of the stress response by the fish. Fish will be stressed by:

- capture in the holding tank for movement to transport;
- the move from tank to transport truck and back to tank;
- poor water quality in the transport tank; and
- high density of fish in the tank.

There are a variety of methods to reduce stress when transporting fish outside of the facility. Reducing water temperature, adding supplemental oxygen to maintain concentrations at or above saturation, adding salt (0.5%), taking the fish off feed 24 to 48 hours before transport, changing the water between multiple trips, and using anesthetics such as MS-222 are all ways that may reduce stress during transport. Note that if MS-222 is used, the fish should probably be taken off feed for more than the minimum 24 hours prior to transport, as the fish will be more likely to defecate and reduce water quality following exposure. If you are hauling fish that will be sold immediately for human consumption, then restrictions apply for withdrawal periods—check with local state authorities. There are studies that indicate that MS-222 itself may stress the fish during its initial exposure to the anesthetic. Since crowding, netting, and transport is where much of the stress response is triggered, putting them in water with anesthetic only attempts to quiet fish that have already been stressed.

Humane treatment of animals is a growing concern among many people. For those shipping live fish, this should be a consideration. While research has been carried out on the effects of stress upon fish, it is incumbent upon the hauler to find ways to keep the animals in the best condition possible. It may be possible to move them through water to the hauling tank rather than using heavy dip nets and carrying the fish to the haul tanks. In this, or by using other techniques designed to minimize stress on the fish during the shipping process, the goal should be to provide the animals with the least stress possible and to arrive at the destination with fish in the best of health.

Even if only a few fish are being transported, always fill the transport tank as high as practical to avoid water movement. Sloshing can result in what is known as "free-surface effect" and can result in fish being damaged by hitting the sides and top of the live-haul tank, as well as loss of control of the transport vehicle as water volume quickly shifts from one side to another. There have been instances of vehicles overturning from this effect.

A summary of important techniques used to reduce fish transport stress are:

- Replace the hauling tank water with fresh water between trips
- Always clean tanks between fills with sanitizing agent
- Maintain oxygen between 100 and 150% saturation
- Add a salt solution to each tank prior to adding fish (5-9 ppt)
- Take fish off feed 24–48 hours prior to transport
- Consider using 15 mg/L MS-222 to sedate fingerlings (restrictions apply to food-sized fish)

EQUIPMENT

Tanks

Hauling tanks have changed considerably over the years. Years ago, tanks were constructed of wood, iron, and steel. They were heavy, often leaked quite a bit, and were hard to sanitize. Today, most modern hauling tanks are constructed of either fiberglass or aluminum. Properly constructed with foam insulation, these tanks can safely haul fish for long distances while minimizing temperature changes. These materials also lend themselves to proper sanitizing between uses to ensure that disease organisms are kept under control, an important factor in producing quality fish.

While tanks can often be built locally, the professional fish farmer would do well to consider commercial companies that have a good track record of building these units. Frequently, they have established designs

and the experience to build tanks that can prevent many problems encountered by those who have had tanks built by people with little fish hauling experience. Tanks ranging is size from 200 liters (50 gallons) to several thousand liters in size are available, although the most common ones hold from 380 liters (100 gallons) to 1,136 liters (300 gallons) of water. Multiple tank units are quite common, combining several individual tanks into one hauling system. This allows for hauling partial loads, multiple sizes, or species, or for several delivery points to be included in a single haul.

Key features of good hauling tanks are:

- Durable
- Easy to repair
- Made of non-toxic materials
- Easy to sanitize
- Insulated
- Watertight
- Systems for dissolved gas control for hauling distances
- Easy to load and unload fish

Durability is a key feature for a tank since it will take a lot of abuse during its life. Aluminum and fiberglass are both good materials to use, although aluminum will normally stand more abuse. The material should be resistant to degradation by ultraviolet (UV) radiation coming from sunlight, which will make some materials brittle and subject to cracking within a year or two. Good hauling tanks should be constructed using foam as an intermediate insulation layer, which further helps stabilize the temperature within the hauling unit. While sturdily constructed units will have little need for repair, sometimes damage does occur. When it does, fiberglass can be more easily repaired without specialized equipment or skills than can aluminum, which requires special welding techniques. The use of non-toxic materials is very important not only because we do not want fish to die, but also because we are dealing in most cases with food products and that should always be a priority in our business.

Sanitizing the hauling equipment is very important and should be considered a normal and regular part of maintenance. Sanitizing agents that wipe out disease causing organisms and keep the transport units clean should always be used at the conclusion of a delivery and before the tanks are used for another run. Preferably, this process should occur "off-site", e.g., a car wash or a location away from the culture building where wash water can be contained and disposed of properly and legally. Biosecurity in the operation should be paramount in order to prevent

disease organisms from becoming established in the system or within the fish populations (see Chapter 11).

Tanks should be sized properly for the intended loads as well as the truck or other vehicle that will carry them, Fig. 15.6. Remember, the weight of water is approximately 1,000 kg/m^3 (8.34 lbs/gal). For a tank holding 1,890 liters (500 gal) of water then, you must figure the weight of the water (1,890 kg or 4,170 lbs) plus the weight of the tank and related gear in order to know what the load on the vehicle will be. In most cases, small pickup trucks will not be useful in live hauling any significant amount of fish because of the weight involved.

Figure 15.6 Two sizes of live haul tanks for a small pick-up and commercial carrier.

The shape of the hauling tank is another consideration. Most tanks are built to get the most volume from the hauling unit. Therefore, they are square or rectangular in shape. However, other shapes have been used and may be more efficient to work with. Some producers have used circular tanks. Other fish hauling units are elliptical, with partially rounded bottoms. This helps to provide mixing of the water for circulation, and the lack of sharp corners keeps fish from being injured. The shape tends to concentrate weight over the center of gravity of the hauling vehicle, which can help to stabilize the ride, and the elliptical areas on the sides can be filled with expanded foam to further insulate the tank from road heat. These shaped tanks are also easy to unload. The drawback to these units is that volume and area will be lost. A circular tank, for instance, may lose over 20% of the area that could be covered with rectangular or square tanks designed to fit in the same area. If maximum capacity for fish is important, this could be a critical consideration. The fish farmer should consider all alternatives and investigate the available options before making a final decision.

When constructing or purchasing tanks, thought needs to be given not only to the size of the tank but to the fittings and options available

that will make it efficient. Proper construction of the tank can help in getting fish loaded and unloaded quickly, thereby keeping them at lower stress levels. Large hatches in the top of the hauling unit should be hinged and provide space sufficient to quickly place fish into the hauling tanks. These hatches should be equipped with dogs or clamps to keep the doors sealed when the vehicle is underway. Gaskets that provide watertight seals are frequently added to ensure that no water is lost when in motion. Most hauling tanks are constructed with several chambers. These may be built either longitudinally or crossways into the tank. These chambers prevent the "free surface effect" of large volumes of water moving from side to side and front to back during the operation of a vehicle. It is possible for this motion to cause the vehicle to become unstable if not checked. This is why the smaller compartments are built to minimize this motion. Also, the higher the water level is, the less the free surface effect will be in the tank.

The tank should have unloading or dump gates that allow it to be emptied quickly. Fish frequently swim against currents, so there may be few fish exiting at the beginning of offloading while many fish may still remain in small puddles of water near the end of the dump. Some tanks are built with sloping bottoms so that they funnel the fish down and out when unloading, however this configuration also uses some of the volume of the tank and may not be efficient when maximum capacity is desired. If round drains with exterior plugs are used in the tank, there should also be a gate inside the tank so that a controlled discharge can be maintained when the entire tank is not being dumped at one time. Also, the drain should be flush with the bottom of the tank so all the water and fish can be released. If a lip exists on the drain, the unit will be hard to completely empty and fish will likely end up remaining in the tank, making disinfection of the tank difficult as well. Make sure that the ability to completely drain a tank compartment exists, even if a small additional floor drain is added and only used for water drainage at the end of the job. Remember, sanitation is paramount.

Dissolved Gas Control

Delivery of dissolved oxygen and removal of carbon dioxide (CO_2) must be carried out through the hauling process for optimum life support. Oxygen can be delivered by air stones or grates that are placed on the bottom of the tank and which release oxygen in streams of small bubbles from a supply tank. Agitators can be provided to help bring oxygen into the water and assist in driving off CO_2. These need to be planned to ensure that they are properly located in the tank unit for maximum effect. 12-volt motors drive the agitators used on most hauling units. These units

need to be considered in sizing the charging system for the vehicle since multiple agitators may require that a larger alternator be included on the delivery vehicle that is purchased. Also, in order to keep the agitators/aerators in operation, the vehicle will have to remain running so that they do not draw down the battery of the vehicle. In some instances, dual battery installations are recommended. Vents are frequently provided at the top of hauling tanks so the gases collecting in the air space at the top of the tank can be exhausted to release the buildup of CO_2 during the haul.

In most, the transport vehicle will be equipped with compressed oxygen cylinders that provide the oxygen needed during transport. Oxygen gas is available in industrial, medical, and aviation grades. Of these, the latter two are more expensive and are not required for use in transporting fish. Industrial grade oxygen works well and is much less expensive than the other two. Oxygen cylinders may weigh up to 68 kg (150 pounds) each and must be firmly secured in the body of the hauling unit to prevent them from falling over or moving during the trip.

Flow meters should be built into the delivery system so that flow can be regulated and monitored closely. Always assume that every piece of equipment in the oxygen delivery system may fail during the trip, which necessitates having backup or replacement parts on board at all times. Fish die quickly without oxygen. Be prepared for the worst-case scenario at all times. Non functioning pressure regulators are frequent points of failure that tend to release all oxygen about half way into your trip.

Another device with much more capacity than gas cylinders are liquid oxygen (LOX) dewars. These are insulated containers are used to contain the liquid oxygen, but the cost of the oxygen is less expensive when compared per cubic unit of oxygen delivered. LOX dewars are also heavier, weighing around 355 kg (780 pounds) each, but a 160 liter container of LOX contains the equivalent of 127,426 liters or 127 m^3 of oxygen (4,500 cubic feet) of oxygen. While compressed oxygen can be stored indefinitely, a LOX dewar will lose about 2 percent daily due to off-gasing.

It must be noted that liquid oxygen can also present a hazard in the event of a vehicle accident. It is kept at a very low temperature since it has a boiling point of -150°C (-238°F). Contacting the liquid directly can cause severe burns to human flesh, in addition to the support for combustion that it can provide in the event of a fire.

In general, if you are operating a smaller truck and making infrequent or short-haul deliveries, you should probably opt for oxygen gas cylinders since they will provide adequate service and are able to be stored for long periods without losing their gas. For long-haul transport, or for operations that are on the road frequently, LOX would be much more economical and advantageous. Always remember that state regulations may apply to hauling gas cylinders and dewars and you should check with authorities in the states that will be transited regarding their use, prior to your first haul.

VEHICLES

In order to successfully transport fish, the size of the hauling unit must be matched to the vehicle. Remember, you are going to be moving a lot of water, which is heavy. In order to properly size a hauling unit, you will need to calculate the amount of fish that will normally be moved. You will have to size the hauling tank for that amount of weight. Then, considering the weight of the water, fish, hauling unit, and related equipment and gear that will be moved, a proper size and type of hauling vehicle can be selected. Consultation with sales personnel at reputable truck dealerships can help calculate the size of truck required to haul the intended loads. Factors such as gross vehicle weight (GVW) rating, engine and transmission sizing, cooling and charging systems, and operator comfort options such as air conditioning and seating should be considered. If the vehicle exceeds certain limits, different classes of operator licenses may be required as well. These should be known before the purchase is made.

Fish hauling is carried out using everything from pickup trucks to large eighteen wheeled tractor-trailer combinations, depending upon the load and distance that must be traveled, Fig. 15.6. The most commonly used version is a straight body truck with hauling tanks that dump to the side. Half-ton pickups can handle tanks up to around 0.4 m^3 (100 gallon) capacity, while larger ¾ and 1 ton versions can be used for trailers with capacities of up to 2.8 m^3 (750 gallons) in two or more units.

The use of trailers that can be towed with a pickup truck is a popular option in fish hauling. Excellent options are available that use a gooseneck or "fifth wheel" trailer. This not only allows a suitably sized trailer to be pulled by the pickup when needed to move a heavy load of fish, but also retains use of the vehicle for general farm use with the trailer disconnected when not on live haul duty.

These types of trailers also place the tanks lower to the ground, which can greatly aid in loading and unloading the fish. Placing the tanks with their loads lower to the ground also helps lower the center of gravity, which increases stability during hauling. Access to the oxygen tanks and related equipment is easier as well, and with the use of the gooseneck trailer, the vehicle can negotiate places where straight body trucks may have difficulty going.

OTHER EQUIPMENT

Monitoring equipment is necessary on hauling rigs to ensure measurement of water quality before and during the hauling process, as well as at the delivery site. The development of electronic meters for measuring dissolved oxygen, pH, and salinity, that are durable and moderately priced has made them a necessity for anyone engaged in hauling as part of their business (Fig. 15.7). Dissolved oxygen meters priced in the $500–750 range are generally very accurate and rugged enough to withstand the rigors of field use. Digital pH meters that provide accurate measurements are now available for under $100. Many of these can fit in a shirt pocket. Salinity can be measured either with a hydrometer (less than $50), a test kit ($50–60), or a meter, any of which can provide measurements that will be accurate enough for hauling. For long-distance transport, consider meters for DO, pH, and salinity, with backup test kits in case of failure. DO and salinity meters will also provide temperature measurement, another requirement for healthy fish hauling.

Fittings used in hauling units should be rustproof, with stainless steel, aluminum, or thermoplastic being the most common. Durability is a key feature of anything used in the business and long life should be sought in all features of the hauling unit.

Regulation of bottled oxygen is carried out using a pressure reducing regulator that can drop the pressure of the tank down to one usable in the hauling unit, usually about 0.7 to 1.0 atmospheres (10–15 psi). Plastic tubing, usually 10 mm (3/8 inch) diameter, is used to carry the oxygen from the regulator to flow meters placed at each tank unit. These regulate

flow in liters per minute to the air stones or diffusion grates. A single oxygen cylinder holds around 7,930 liters (280 cubic feet) or 10.4 kg (23 lb) of gas. Calculation of depletion of oxygen gas can be made based on how much oxygen will be flowing and the duration of the trip. Agitators are used to remove excessive CO_2 and to provide oxygen in low density loads (see Fig. 15.8).

Figure 15.7 Monitoring equipment for live hauling of fish (clockwise from top: waterproof pH meter, oxygen meter with cable and probe, digital thermometer).

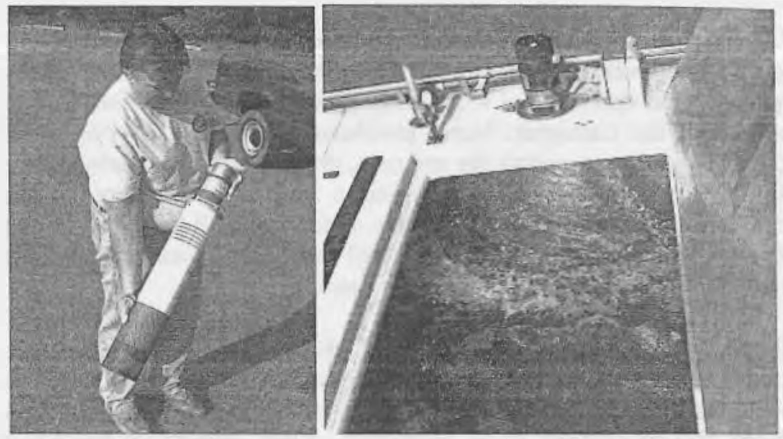


Figure 15.8 Agitators used in live haul tanks (Don Webster, University of Maryland, holding agitator).

PERMITS

A very important consideration that should never be left out of the hauling operation is permitting. Before moving fish either within or between states, the operator should make sure that correct permits have been obtained for the fish being moved. Additionally, the hauler must have the proper paperwork in possession, in conformance with the requirements of the territory the fish are moving across. This may involve calling natural resource management agencies in the states that will be transited to ensure that tagging and paperwork are in order. In addition, some states have regulations on markings that must be on fish hauling trucks. Remember that crossing state lines with fish that may be illegal in another state can not only render you liable to severe penalties and also subject you to prosecution under Federal law. This is serious business. Do not underestimate the vigor that authorities will use to pursue you. Some fish farmers have been put in prison for violating these rules. Be careful. If you have any doubts, check with your local state authorities and obtain all answers in writing.

WATER QUALITY

In order to deliver healthy fingerlings to the culture facility, or to have quality live product arrive at the destination, fish must be treated properly throughout the hauling process. While catastrophic losses will occur during the loss of life support systems, such as loss of oxygen, most hauling losses are more subtle. They occur through disease that is

induced by stressing fish during the processes of loading, hauling, and delivery. The effects of these stressors may not manifest themselves for several days after delivery but will almost certainly lead to dissatisfaction on the part of the customer. Many problems are related to poor water quality, improperly tempering the animals, or overcrowding them during hauling. Several water quality parameters must be considered when hauling. Since most of these are similar to factors in RAS systems and are covered elsewhere in this book, we will limit discussion here to those factors specifically related to proper fish hauling.

Dissolved oxygen (DO) is critical for life support of your fish. Maintain DO levels at 6 mg/L in the hauling unit. If well water is used to fill the tanks, this must usually be oxygenated before the fish are added to ensure that the water is at saturation. Since saturation occurs at different levels depending upon the temperature of the water and salinity, check the chart in the Appendix to find the level that should be available. DO is introduced into the water through air stones or other delivery systems. Tubing is available that provides small bubbles that disperse oxygen gas into the water. Generally, the smaller the bubbles, the more effectively they will be at providing dissolved oxygen to the water. Oxygen meters can be designed to measure DO levels in the tanks during the hauling process and can be read in the cab while the vehicle is moving, or DO can be read at planned stops. Remember that fish can die in as little as 15 minutes if an oxygen delivery component fails. Even if the fish do not die immediately, anoxia (low DO) may stress them to the point where disease will set in within a day or two and kill them later.

Carbon dioxide (CO_2) is a byproduct of respiration and must be removed to ensure that the fish remain healthy. A buildup of CO_2 can prevent the blood of the fish from effectively carrying oxygen. Agitators are normally used to dissipate CO_2 in the hauling unit, although electric bilge pumps that spray the hauling water up against the top of the tank are also used. Keep in mind that this is only effective if there is gas exchange or venting from the tank unit to disperse the CO_2; otherwise the CO_2 will dissolve into the water column again. Think in terms of moving 5 to 10 volumes of air through the purging air space for every unit of water that is pumped or moved by the surface agitator.

Ammonia is part of the nitrogen cycle and enters hauling water as excretion products from the fish. It can be toxic to fish in water having a medium to high pH (pH >7.5) and low levels of dissolved oxygen, as it decreases the ability of the fish to take oxygen into the bloodstream and can cause anoxia, or suffocation, of the animal. Ammonia is easily measured with test kits that compare the developed test sample against a standard for a reading. Levels as low as 0.06 mg/L of un-ionized ammonia-nitrogen can damage gills and impair the health of normal fish. Remember: taking fish off their feed for 24 to 48 hours prior to transport helps reduce ammonia production to nearly zero. In most cases, a pH range of 6.5 to 8.0 is suitable for hauling, although the closer to neutral the water can maintained the better. pH is one of the factors that must be known in the source water, the hauling water, and the receiving water and matched as closely as possible to ensure that the fish are kept in the same ranges. Consulting with the customer receiving the fish should include asking what the receiving water pH is. Alkalinity helps maintain a neutral pH through buffering and levels above 20 mg/L are considered beneficial. The ammonia disassociation is pH dependent (see Chapter 2 Water Quality) with un-ionized, or "free" ammonia, increasing as pH increases. Therefore, if ammonia levels are expected to be high, you might make an attempt to keep water pH below 7.0 so there is less un-ionized ammonia (NH_3, toxic form) that will harm fish. (Note a pH of 7.0 will have 50% of the un-ionized free ammonia of water at a pH of 7.3.)

Alkalinity levels of 50 to 100 mg/L are considered very good for fish, while levels below 20 mg/L are considered soft and not favorable to them. Alkalinity and hardness can be increased through the addition of sodium bicarbonate and calcium chloride, respectively. To increase alkalinity by 10 mg/L, add 13 g per m3 (5 grams per 100 gallon) of sodium bicarbonate of the hauling water. To increase hardness by 10 mg/L, add 15.1 g per m3 (6 grams per 100 gallons) of calcium chloride.

"Rule of Thumb"

To increase alkalinity by 10 mg/L:
Add 6.3 g sodium bicarb per 379 L (100 gallon)
or 16.6 g per m^3

To increase hardness by 10 mg/L:
Add 5.7 g calcium chloride per 379 L (100 gallon)
or 15.1 g per m^3

Salinity is a consideration in hauling operations in several ways. First, fish raised in salt or brackish water should always have the hauling water matched to the salinity that they came from. For other species, the addition of salt to hauling water is a widely used practice that aids the osmotic balance of the fish. Salt is generally added from 0.5 to 1.0

percent (5 to 10 ppt), depending upon the species. Salinity levels can be easily and inexpensively measured by using a hydrometer calibrated for salt. These are available at most pet stores dealing in fish. More sophisticated units are available as meters and are urged for professional fish haulers.

TEMPERATURE AND TEMPERING

Temperature is an important consideration in hauling since there are minimum and maximum temperatures at which fish can live. Also, warmer water has lower oxygen solubility, and fish will be more active due to higher metabolism, which will result in higher oxygen demand by the animals, while at the same time having less oxygen available because of the lower oxygen solubility level. The hauler should be aware that sudden changes in temperature are likely to be harmful to fish. While fish can be taken on longer hauls and at greater densities by reducing the temperature of the hauling water, this must be done gradually to avoid thermal shock. Similarly, when the fish arrive at their destination, the temperatures of the hauling and receiving waters may take some time to equalize so that effective transfer can take place.

Tempering is the process in which fish are slowly acclimated to the hauling water or the receiving waters. It must be stressed that this needs to be done slowly and that sufficient time must be taken to properly match water conditions in order to transfer fish that will remain healthy and robust. The lethal temperatures for the species must be known so that they are not exceeded. For example, trout (various salmonids) should be hauled in water that is about 11 to 14°C (52–57°F), while tilapia will die in water that cold. Tilapia would need to be kept in water that is in the 21 to 26°C (70–78°F) range.

Temperature differences of more than 5.5°C (10°F) can harm fish and smaller ones are more susceptible to these changes. To lower water temperature, ice is frequently used because it is readily available and additional supplies can either be stored in insulated compartments or purchased along the route. Ice should be made from de-chlorinated water since some species are sensitive to this chemical. The addition of ice to the hauling water will lower the temperature and metabolism of the fish while raising the oxygen saturation point of the water. One pound (0.45 kg) of ice will usually lower two gallons (7.6 kg or 7.6 L) of water 5.5°C (10°F) (see Table 15.1 for amount of ice needed to cool water volumes). Temperature changes should be made gradually so that the fish are not thermally shocked. This change should not exceed 5.5°C (10°F) in any 20 minute period.

When hauling fish for short distances in insulated tanks, the temperature of the shipment water can be matched to that of the destination receiving water at the start of the trip. In this way, fish can be offloaded as soon as the vehicle arrives at the destination preventing costly holding time for the driver and the unit. For longer distances and trips that will take from eight to twelve hours or more, the buildup of temperature can take place beginning several hours before arrival. Proper planning can help to ensure that deliveries move as quickly as possible while maintaining the fish in top condition. Experience is the best teacher in estimating the change in temperature at a given time of year. This is also why experienced drivers are highly prized by employers.

Table 15.1.a. Required (approximate in kg) Mass of Ice to Cool Water

Volume (Liters)	Desired Change in Water Temperature[a] 2.5° C	5° C	7.5° C	10° C
200	6.0	12	18	24
300	9.0	18	27	36
400	12	24	36	48
600	18	36	54	72
800	24	48	72	96
1,000	30	60	90	120
1,500	45	90	135	180
2,000	60	120	180	240
2,500	75	150	225	300
3,000	90	180	270	360
4,000	120	240	360	480

[a]NOTE: fish should be tempered at least 20 minutes for EACH 5° C change in water temperature

Table 15.1.b. Required (approximate) Pounds of Ice to Cool Water

Volume (Gals)	Desired Change in Water Temperature[a] 5°F	10°F	15°F	20°F
50	12	25	38	50
100	25	50	75	100
200	50	100	150	200
300	75	150	225	300
400	100	200	300	400
500	125	250	375	500
600	150	300	450	600
700	175	350	525	700
800	200	400	600	800
900	225	450	675	900
1,000	250	500	750	1,000

[a]NOTE: fish should be tempered at least 20 minutes for EACH 10°F change in water temperature

Consider pH when moving fish since they can be sensitive to extreme changes. When arriving at the destination, a reading should be made of the pH of the receiving water. If there is more than 1 unit difference, the receiving water should be slowly pumped into the hauling tank so that the fish can acclimate properly. Replacing ten percent of the tank water every 10 to 20 minutes until the pH readings are stable will prevent fish from undergoing shock upon transfer.

Loading

Only healthy fish should be shipped. They should be taken off feed from one to three days prior to shipment, depending upon the water temperature in the culture unit. With higher temperatures, the metabolism of the fish will increase and the fish will utilize their feed faster than in colder waters, when additional time will be necessary for them to empty their gastro intestinal system. Taking the fish off feed will allow many of the waste products to be purged that would otherwise be excreted into the hauling water or regurgitated during handling and shipment. This will affect the hauling water and cause water quality problems to occur. Fish tend to become excited during transfer, especially when they are netted or crowded, and can develop sores or lesions if not handled quickly and properly. When moving fish with dip nets, care should be taken to use netting that is soft and knotless,

especially on species that have scales. These scales protect against disease and, when rubbed off, can allow fungal and other disease organisms to rapidly develop, yielding an unsightly and inferior product for the live market.

Loading should occur in low light conditions and, preferably, not at the warmest time of the day. During windy conditions, fish can dry out and, if temperatures are low, wind chill can have a profound effect upon them and leave many dead in a short time. This is especially true of warm water species such as tilapia when moved during winter. Loading rates for hauling fish depend upon several factors:

- Species to be moved
- Size of the fish
- Duration of the haul
- Water temperature

"Rule of Thumb"

New haulers should reduce capacities by 33% to 50% of maximum; then increase load levels as experience and confidence are gained.

These factors assume that the fish are in good condition and that the hauling vehicle is adequately designed to provide the necessary life support such as oxygen input, removal of CO_2, minimal buildup of ammonia, with pH, alkalinity, and hardness within proper ranges.

The hauler should be cautioned to start with a known acceptable loading rate and only through experience should this rate be adjusted. Some species can sustain higher loading rates than others. Fewer small fish can be hauled than larger ones and the warmer the water is, the lower the hauling density should be. Also, the greater the hauling time, the lower the density should be.

Loading rates have been calculated for many species such as trout and catfish but there are still gaps for species such as tilapia and yellow perch; see Tables 15.2–4 for specific fish species and use Table 15.5 as a general guide for allowable hauling rates. Adjustment should be made based on the temperature of the water and whether or not efficient oxygen transfer systems are used in the hauling unit. As always, the prudent culturist will begin with a conservative loading rate and increase it only with experience.

Table 15.2.a. Loading Rates (kg per liter) for Catfish in 18.3°C Water

Size of Fish (kg per 1.000 fish)	Hauling Time (hrs) 8	12	16
.045	0.024	0.024	0.024
.45	0.150	0.120	0.084
.91	0.210	0.198	0.150
1.8	0.264	0.210	0.180
3.6	0.353	0.264	0.216
9.1	0.413	0.300	0.246
113	0.599	0.491	0.353
227	0.707	0.575	0.413
454	0.755	0.665	0.575

NOTES: 1) loading can be increased 25% for each 5.6°C drop in temperature below 18.3°C.
2) loading should decrease 25% for each 5.6°C increase in temperature above 18.3°C.

Table 15.2.b. Loading Rates (lbs per gallon) for Catfish in 65°F Water

Size of Fish (lbs per 1,000 fish)	Hauling Time (hrs) 8	12	16
0.1	0.20	0.20	0.20
1	1.25	1.00	0.70
2	1.75	1.65	1.25
4	2.20	1.75	1.50
8	2.95	2.20	1.80
20	3.45	2.50	2.05
250	5.00	4.10	2.95
500	5.90	4.80	3.45
1,000	6.30	5.55	4.80

NOTES: 1) loading can be increased 25% for each 10°F drop in temperature below 65°F.
2) loading should decrease 25% for each 10°F increase in temperature above 65°F.

Table 15.3.a. Loading Rates for Warmwater Fish (Largemouth Bass, Bluegills, and Tilapia) in Water of 18 C° for Hauls of Less Than 30 Hours Duration

Length (cm)	Number Fish per kg	Approx. Number per liter	Loading Rate (kg per liter)
12.7	22	4	0.18
10.2	55	7	0.13
7.6	220	18	0.08
5.1	880	53	0.06
2.5	2200	89	0.04

Table 15.3.b. Loading Rates for Warmwater Fish (Largemouth Bass, Bluegills, and Tilapia) in Water of 18 C° (65°F) for Hauls of Less Than 30 Hours Duration

Length (inches)	Number Fish per pound	Approx. Number per gallon	Loading Rate (lbs per gal)
5	10	15	1.50
4	25	25	1.00
3	100	67	0.66
2	400	200	0.50
1	1,000	333	0.33

Table 15.4.a. Loading Rates for Coolwater Fish (Walleye and Northern Pike) in Water of 12-18 C°

Number per kg	Size cm	kg of Fish per liter	Transit Time (hrs)
132	7.6	0.34	8.0
1100	5.1	0.17	8.0
2200	2.5	0.14	8.0

Table 15.4.b. Loading Rates for Coolwater Fish (Walleye and Northern Pike) in Water of 55–65°F

Number per pound	Size (in)	Lbs. of Fish per gal	Transit Time (hrs)
60	3.0	1.30	8.0
500	2.0	0.66	8.0
1,000	1.0	0.55	8.0

Table 15.5.a. General Guidelines for Loading (kg per liter) of Various Types of Fish at 18 C°

Type and Length Of Fish (cm)	Duration of transport (hrs) 1	6	12	24
Fingerling food fish				
5.1	0.24	0.18	0.12	0.12
20.3	0.36	0.36	0.24	0.18
Adult food fish				
35.6	0.48	0.48	0.36	.24
Baitfish				
5.1	0.24	0.18	0.12	0.12
7.6	0.36	0.24	0.12	0.12

Table 15.5.b. General Guidelines for Loading (lb per Gallon) of Various Types of Fish at 65°F

Type and Length Of Fish (inches)	Duration of transport (hrs) 1	6	12	24
Fingerling food fish				
2	2.0	1.5	1.0	1.0
8	3.0	3.0	2.0	1.5
Adult food fish				
14	4.0	4.0	3.0	2.0
Baitfish				
2	2.0	1.5	1.0	1.0
3	3.0	2.0	1.0	1.0

We can calculate the loading density of a tank using the water-displacement method. For this, we need to know:

- Actual volume of the tank being used;
- Weight of the fish that will be transported; and
- Volume of the water that will be displaced by the fish (see Table 15.6).

Table 15.6.a. Volume of Water Displaced by Fish Added to Hauling Vessel

Weight of Fish (kg)	Water Displaced (liters)	Weight of Fish (kg)	Water Displaced (liters)	Weight of Fish (kg)	Water Displaced (liters)
45	45	680	681	1270	1272
90	91	726	727	1315	1317
136	136	771	772	1361	1363
181	182	816	818	1406	1408
227	227	862	863	1451	1454
272	273	907	908	1497	1499
318	318	953	954	1542	1544
363	363	998	999	1588	1589
408	409	1043	1045	1366	1635
454	454	1089	1090	1678	1681
499	500	1134	1136	1724	1726
544	545	1179	1181	1769	1772
590	591	1225	1226	1814	1817
635	636				

Table 15.6.b. Volume of Water Displaced by Fish Added to Hauling Vessel

Weight of Fish (Lb)	Water Displaced (gal)	Weight of Fish (lb)	Water Displaced (gal)	Weight of Fish (lb)	Water Displaced (gal)
100	12	1500	180	2800	336
200	24	1600	192	2900	348
300	36	1700	204	3000	360
400	48	1800	216	3100	372
500	60	1900	228	3200	384
600	72	2000	240	3300	396
700	84	2100	252	3400	408
800	96	2200	264	3500	420
900	108	2300	276	3600	432
1000	120	2400	288	3700	444
1100	132	2500	300	3800	456
1200	144	2600	312	3900	468
1300	156	2700	324	4000	480
1400	168	2800	348	4100	492

From these, loading density can be calculated:

$$Loading\ Density\ (kg/L) = \frac{Kg\ of\ fish}{TankCapacity(L) - Water\ displaced\ by\ fish(L)}$$

or

$$Loading\ Density\ (lb/gal) = \frac{pounds\ of\ fish}{tank\ capacity(gal) - water\ displaced\ by\ fish(gal)}$$

Example 1: We are going to place 408 kilograms of fish in a 2271 liter tank, we find from Table 15.6.a. that water displaced will be 409 liters. From our formula we calculate:

$$Loading\ Density\ (kg/L) = \frac{408Kg\ of\ fish}{2271(L) - 409(L)} = 0.22kg/L$$

Example 2: We are going to place 900 pounds of fish in a 600 gallon tank, we find from Table 15.6.b. that the water displaced will be 108 gallons. From our formula we calculate:

$$Loading\ Density = \frac{900lb}{(600 - 108)gal} = 1.83\ lb/gal$$

ADDITIVES

When producing food fish, the aquaculturist always needs to remember that the product is human food and, as such, should be treated in a way that ensures that it is safe and wholesome. Chemicals and additives used in production come under the control of the US Food and Drug Administration (FDA) and are part of a stringent set of laws enacted by Congress. Any substance used on fish to be used as human food comes under the auspices of the FDA. Some, such as salt and ice have been deemed to be "Generally Regarded as Safe" (GRAS) and are allowed to be used until such time as they may be objected to. For hauling purposes, ice, salt, and oxygen used to move fish are allowed but anesthetics are not. There is a 21-day waiting period before fish that have been treated with anesthetic can be sold for human consumption. In the normal conduct of business, this makes them impractical for shipments destined to market. See Table 15.1 for ice addition considerations and 15.7 for calculations on the use of salt.

Fingerlings have been transported with anesthetic with great success. Used according to the label, anesthetics such as methane tricainesulfonate (MS-222) are able to reduce the metabolism of fish, thereby lowering their oxygen consumption and allowing them to be

transported successfully over longer distances than might otherwise be possible. This is another factor that should be considered when transporting fish, however, since it may also add to the time required to temper the fish when they have arrived at the destination in restoring them to their normal state.

Table 15.7.a. Required Salt Addition (in kg) to Achieve a Specific Salinity Level in the Hauling Tank

Volume	Salinity in Parts Per Thousand (ppt)							
(liters)	1	5	10	15	20	25	30	35
50	.05	.25	0.5	.75	1.0	1.2	1.5	1.8
100	0.1	0.5	1.0	1.5	2.0	2.5	3.0	3.5
200	0.2	1.0	2.0	3.0	4.0	5.0	6.0	7.0
300	0.3	1.5	3.0	4.5	6.0	7.5	9.0	10.5
400	0.4	2.0	4.0	6.0	8.0	10.0	12.0	14.0
500	0.5	2.5	5.0	7.5	10.0	12.5	15.0	17.5
600	0.6	3.0	6.0	9.0	12.0	15.0	18.0	21.0
700	0.7	3.5	7.0	10.5	14.0	17.5	21.0	24.5
800	0.8	4.0	8.0	12.0	16.0	20.0	24.0	28.0
900	0.9	4.5	9.0	13.5	18.0	22.5	27.0	31.5
1,000	1.0	5.0	10.0	15.0	20.0	25.0	30.0	35.0

Table 15.7.b. Required Salt Addition (in lbs.) to Achieve a Specific Salinity Level in the Hauling Tank (Note: 1 gallon = 3.785 L and 264 gallons = 1 m^3)

Volume	Salinity in Parts Per Thousand (ppt)							
(Gals)	1	5	10	15	20	25	30	35
50	0.4	2	4	6	8	10	12	14
100	0.8	4	8	13	17	21	25	29
200	1.6	8	17	25	33	42	50	58
300	2.6	13	25	38	50	63	75	88
400	3.4	17	33	50	67	83	100	117
500	4.2	21	42	63	83	104	125	146
600	5.0	25	50	75	100	125	150	175
700	5.8	29	58	88	117	146	175	204
800	6.6	33	67	100	133	167	200	233
900	7.6	38	75	113	150	188	225	263
1,000	8.4	42	83	125	167	208	250	292

There are commercial formulations of additives that may be used for hauling and are approved for use on food fish. Some of these provide buffers for the water so that the pH and alkalinity may be optimized during hauling. Others provide anti-foaming agents for the hauling tanks so that protein foam does not build up during the moving process. Foam can prevent proper ventilation of the tanks, allowing buildup of carbon dioxide due to lack of gas venting. The hauler should always know the intended purpose for the fish being moved and, if they are being used for food, should use nothing that is not specifically approved for use on human-use products.

SANITIZING

Disease can severely affect fish farms. Keeping all equipment as clean and sanitary as possible can prevent costly loss of fish. As part of a comprehensive biosecurity plan, hauling equipment should always be cleaned and sanitized at the conclusion of the transport. Pressure washers provide an effective means of cleaning and ensuring that microbacterial contamination does not occur. Sanitizing agents should be used at all times and the equipment should be well rinsed so that no chemicals remain that could harm future fish cargo.

While regular household bleach may be used as a sanitizing agent, its concentration is only about 5 percent chlorine. HTH is stronger and provides an excellent means of killing potential disease organisms. HTH contains a calcium hypochlorite base with 65 percent of available chlorine. Used at a level of 15 mL per 100 L (½ ounce per 25 gallons) of water, it should be used on all tanks, pumps, lines, and equipment for 30 minutes for proper disinfection. Formalin has also been used as a disinfectant on fish farms at a 5 percent solution. Do not forget to include all nets, boots, gloves, baskets, and other equipment was used on any haul as well. Do not give disease organisms any opportunity to become established in your business. (See Chapter 16 Fish Health management for more details on biosecurity).

15.8 PURGING AND OFF-FLAVOR

One of the problems often encountered in recirculating systems is development of off-flavors in the fish. One method of eliminating the off-flavor is to hold the fish for 3 to 5 days in a tank with fresh, clean water. The water exchange rate, which should be supplied by fresh water, e.g., well water, should be at least 25% volumetric exchanges per day

and preferably 100 to 200% on the first day. If you do not flush out the pollutants from the water, these compounds will not diffuse away from the fish. Withdraw feed at least 24 hours before moving the fish to the purging tank. This will decrease the water exchange requirements in the purging tank. It also decreases the oxygen demand of the fish once they are moved to the purging tank. Off-flavor uptake by fish is rapid and elimination is a slow process.

Fish grown in RAS are very susceptible to off-flavor (muddy taste). There is no more certain way to lose customers than to ship then fish with off-flavor. Before harvesting, take a fish and sample for off-flavor. Off-flavor is primarily attributed to geosmin and methylisoborneol. These are metabolic compounds produced by actinomycetes (Gerber, 1979), cyanobacteria (Slater and Blok, 1983) or algae (Juttner, 1983). Detection of these compounds is at the fractions of a microgram per kg of flesh (Person, 1979). A thorough review of off-flavor issues is provided by Brune and Tomasso (1991).

Testing for off-flavor is usually done by taking a fillet and cooking it in a microwave oven without any seasoning and then having someone who has a taste sensitivity to do the taste test. If there is a "hint" of off-flavor, then delay the sale and continue purging. Consider increasing your percentage of fresh water being added.

15.9 POST HARVEST HANDLING

Production facilities selling dead in-the-round or dressed fish must provide refrigeration facilities for short term storage of the product and should in most cases have an ice machine either to provide for all of the cooling or to supplement any mechanical refrigeration. Dead fish retain quality much better if kept as close to freezing as possible. For example, fish held at -2°C (just above their freeing point) have a shelf life several days longer than fish held at + 2°C. Fish shipped on ice retain better quality, in most cases, than if held under standard refrigeration at the same temperature, because the ice provides high humidity, and a washing action, as well as cold temperatures. Large block ice should be avoided. The weight of the ice can crush fish, sharp edges from cracking the blocks can tear the fish, and the large pieces will make poor contact with the fish slowing the cooling rate. If boxes of fish are to be stacked,

then fish and ice should not be packed beyond the stacking line. Drainage of melted ice water from the fish should be permitted, and ideally not dripped directly onto fish in the box beneath. Fish should be packed so that dirty melt water does not collect in the belly cavity of the fish. Fillets or other "flesh" surfaces should not be in direct contact with the ice.

The impact of temperature on the shelf-life of fish and other products has been carefully studied in Australia. The results of these studies really highlight the importance of keeping fish as cool as possible. They have developed an equation that relates spoilage rate (SR) to the temperature of the fish (whole body or flesh):

$$SR = b(T - T_0)^2 \tag{15.3}$$

where:

T = temperature of the fish °C,
T_o = reference temperature for the fish spoilage process °C,
b = proportionality constant that depends on the shelf-life of the particular fish.

Surprisingly, T_o is −10°C, because microbes can grow essentially down to that temperature. Although the value of "b" can be calculated, the important term to understand is the $(T-T_o)^2$ term and the impact it has on shelf life. The shelf-life is the reciprocal of the spoilage rate for each species, i.e., the faster the SR, the shorter the shelf-life, or the slower the SR, the longer the shelf-life. It should also be noted that the simple equation above only holds in the "refrigeration" region. At higher temperatures the equation becomes more complicated.

With that introduction, let us look at a table of values for the $(T-T_o)^2$ term assuming that the fish of interest is normally considered to have a shelf-life of 14 days (see Table 15.8).

Table 15.8 Relative Spoilage Rates (SR) as Affected by Storage Temperature

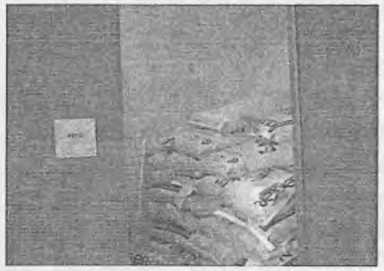
Temperature (°C)	Relative SR $(T-To)^2$	Relative Shelf-life (days)
-2	64	21.8
0	100	14.0
2	144	9.7
4	196	7.1
6	256	5.5
8	324	4.3

As you can see, the impact of small temperature changes is significant. Furthermore, it should be noted that normal "Refrigeration" could legally be defined to go up to 45°F (7°C). Obviously fish kept at that temperature spoil a lot faster! Good "refrigeration" is generally considered to be below about 40°F (4°C), but even this is insufficient for fish (and many other products, as the relative relationship holds). So, for ideal fish storage, temperatures of 33°F or even lower are recommended. Superchilled (unfrozen) fish will have an even longer shelf-life, i.e., fish kept at temperatures between 32°F (0°C) and 28°F (-2°C), often using salt-water ice, which is colder than freshwater ice. Thus, another benefit of ice is the ability to keep fish colder than most available mechanical refrigeration systems.

15.10 Feed Storage

Feed must be available on a daily basis and therefore must be at least temporarily stored on site. The type and size of the culture operation, the species cultured, the frequency of feed delivery from the vendor, and other factors will determine the type and size of feed storage requirements. Typically, feed is available either in bags or in bulk. Where small quantities are needed, bagged feed is most convenient. Larger operations will find bulk feed more convenient and less costly (see outside storage bins in photo; shading or inside barn placement is generally not considered necessary). However, bulk feed requires handling equipment that may not be necessary for bagged feeds. Feed storage facilities are of two general types: 1) dry, rodent resistant facilities, and 2) refrigerated storage. Most commercial fish feeds can be stored in facilities that are dry and rodent resistant for at least reasonable time periods, i.e., a few weeks, depending on temperature and humidity. Storing feeds too long risks loss of required vitamins and possible spoilage. Some feeds must be refrigerated at all times to prevent spoilage. Feed storage facilities need to be

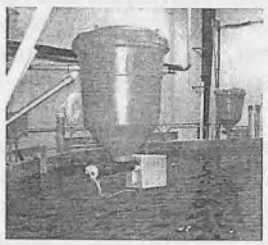
designed to accommodate the feed type and form used. See Chapter 18 for more details on feed degradation.



Under optimal conditions of low temperature and low relative humidity, dry pelleted feeds can be stored 90–100 days. Keep feeds in a room or cooler that is well ventilated and which provides security against rodents and insects. Elevate feed by placing it on pallets and stack feed in bags less than 10 bags high for worker-safety and to prevent crushing the feed at the bottom. High moisture levels and temperatures will rapidly result in poor quality feed. Moisture fosters mold growth and attracts insects. High temperatures break down oils and deteriorate vitamins. If feed becomes rancid or moldy, do not feed it to the fish. The moisture level in pelleted feed after drying will be from 6–8%. Every effort must be made to not let the feed gain moisture once it has arrived at the farm in this dry condition.

It is recommended that samples of the finished products be stored for as long as six months after manufacture, either in refrigerated storage or at ambient temperature. Ambient temperature rather than frozen storage is recommended, so that should concern be raised about a feed, a chemical comparison can be made with feed of the same age that has been stored cool and dry. Complete records should be kept for each batch of feed delivered to farmers. These data should include details such as the date of delivery, batch number, and quantity delivered. Many complaints to the manufacturer about the poor quality of their feed are very often the result of post delivery improper storage conditions on the farm.

Feed Delivery Methods and Delivery Costs



When getting price quotes on feed, also request shipping charges. The shipping charges for the feed can approach the cost of feed. To minimize shipping charges, try to purchase as much feed as possible at once. In addition, purchases of a ton or more often qualify for discounts. Farms using more than 100 tons/yr will generally purchase feed in bulk deliveries of 10 tons or more (a fully loaded truck will haul around 20 ton). Be careful about the way feed is moved from the delivery truck into the bins and from the bins inside the barn to the fish tanks. Each of these processes can increase the fine solids present, which will severely degrade water quality.

In small systems, feeding is easily handled by manual methods or by demand feeders, because the weight of feed moved is small. However, in larger systems many tons of feed will have to be fed. Automated methods of handling this feed are necessary and cost effective. The feed distribution facilities needed, will depend on the management system used in a particular operation. Regardless of the system used, some feed bins will be needed as well as some equipment to distribute the feed. Space and funds to purchase or construct and/or install the equipment will be required.

15.11 Handling Fish

Few, if any, aquaculture facilities are designed to optimize handling of any materials used or produced in the facility. Harvesting usually consists of manually dipping the fish out of the tanks. Manual systems work well if there are only a few thousand pounds of fish produced per year. However, if production is measured in tons per year, handling of the fish becomes a major time and labor consideration, and, thereby, a major cost factor. It is also backbreaking work. Adequate planning during facilities design can save much grief later when handling fish. Equipment must be provided where automated handling is to be used (Fig 15.9). Unfortunately, little research has been done on how best to move fish from one tank to another, when reducing stocking densities. Pumps can be used and they often cause less stress on the fish than do standard dipping methods. How fish handling systems are designed can determine the labor needed for this operation and can determine how much stress the fish will experience.

Figure 15.9 Harvesting artic char with a crowder system (Freshwater Institute).

15.12 LABOR

●ne of the most demanding aspects of aquaculture is the need for labor that is dedicated to the task and has the necessary knowledge and training. When there are fish in the recirculating system, someone has to be present at the facility or on call 24 hours per day, 7 days per week or 24-7. If the operation is small and is operated by one person, that individual must be willing to be on call continuously for long periods of time. The person must be dedicated to the job. Large operations can afford to hire more than one person, so the on-call burden can be spread acr●ss different people. Everyone can get some time off, if all employees take their turn being on-call during some weekends and/or holidays. Particularly in small operations, being continuously on-call requires dedication that few people are willing to make, and is one of the most overlooked aspects of aquaculture production.

"Rule of Thumb"

Being continuously on-call to a fish facility is one of the most overlooked aspects of aquaculture production.

Aquaculture production requires a mixture of knowledge learned in a classical school or college setting, as well as knowledge learned on the job. Currently, there are only a limited number of people who have the needed combination of knowledge and experience to successfully operate a recirculating aquaculture facility. Any sizable recirculating aquaculture operation must make a concerted effort to hire people possessing most of the needed knowledge and to be prepared to train these people on the parts they are missing and to train new people from the beginning of their employment. The manager, owners, and/or investors must be aware that training new people will involve a certain amount of risk and probably will involve loss of at least some crop.

The location of the aquaculture facility will strongly influence the type of people that can be hired. Are the local conditions such that people with the educational backgrounds and experience needed are going to be satisfied to live there? Are the needed people available locally or must they be moved to the site? ●bviously, the availability of a suitable labor supply is not the only determining factor in site location, but it is the one most often overlooked.

15.13 ACCESS

Access to the aquaculture site is necessary for employees, feed supply trucks, oxygen supply trucks, fish haulers, and other functions. This implies that there is a road or some method of getting people and vehicle traffic in and out of the site. There must be available access in all weather conditions and there must be space enough to maneuvers trucks and other vehicles. These areas may be gravel covered or hard surfaced. The physical size of these areas will depend on the type of truck expected and the frequency of truck traffic. The turning radius of a tractor trailer is about 65 feet (20 m), which means that the smallest radius on any driveway should also be no less than 65 feet (20 m). Plan for efficient ingress and egress from the farm site for all large trucks. Loading docks may also be necessary. These facilities can be expensive, particularly if not properly planned into the overall facilities design.

15.14 OPERATIONS

Fish production is more likely to be successful and efficient when good fish rearing habits are employed, when techniques are continuously refined, and when realistic long-range planning is done. Many of the techniques discussed below have been adapted from rainbow trout culture methods and can be used for many other fish species.

FISH SAMPLING

Sampling is used to estimate growth by measuring the weight and/or length of a subset of the entire population. Tracking fish growth accurately is necessary for rationing feed, calculating tank densities, and for projecting the time when fish will be ready for sale. By collecting some of the fish from the tank in question, and comparing the information over multiple sampling intervals, growth rates for the entire population can be estimated.

Growth rates are used to:

- predict when the fish will be ready for future production milestones;
- decide whether past performance has been acceptable; and

- determine when some problem that, while not causing fish mortality, may be present and affecting growth.

By analyzing growth rates for different cohorts of fish in conjunction with other production data, e.g., feeding information and water quality conditions, the manager can determine how production efficiencies change from cohort to cohort and how production parameters will affect the fish, and make adjustments to optimize fish production.

PLANNING

Planning the sampling method, organizing the equipment, and having the necessary number of personnel to help can greatly reduce the time it takes to sample fish. While it is possible to sample with one person, two people are preferable – one person to handle the fish while the second records information and operates the balance scale. Teamwork improves accuracy and efficiency, and reduces the tedium of sampling. Efficient and proper technique resulting from teamwork minimizes fish stress and damage that can lead to post-sampling mortality.

Sampling is stressful to fish. Decisions about how often to sample must balance the need for accurate numbers with the desire to minimize fish stress. Fish sampling can be carried out as frequently as every two weeks, but generally, producers sample every four weeks.

Fish larger than 10 grams should be taken off feed for 24 hours prior to sampling. Consumption of oxygen by fish increases during sampling. Oxygen levels should be allowed to stabilize after sampling, and fish should not be fed for several hours post-sampling. Frequent sampling will allow for more efficient feed rationing and prediction of growth, but will also create stressful conditions for the fish and necessitates taking the fish off feed. Activities that disrupt feeding for more than one day, such as grading, sampling, and transporting fish, should be carried out as infrequently as possible. In the case of rainbow trout, fish can reasonably compensate for one day off feed by eating more food the next day, however, they cannot compensate for two consecutive days off feed per week.

A monthly sampling interval is a reasonable compromise between minimizing stress and maximizing accuracy. Be aware of environmental stressors that may affect the fish when they are removed from the tank for sampling. For example, if the building air has large temperature fluctuations during the day, consider sampling at a time when the air temperature is closest to the water temperature. The bucket used for weighing sampled fish should have a diameter at least as large as the

fish. The fish should be emptied out of the bucket before they start showing adverse signs of stress, including piping or air gulping and excessive thrashing. The amount of water placed in the bucket and the number of fish you can fit in the bucket before emptying it is subjective, and can be determined with a dissolved oxygen meter and some common sense.

SAMPLING PROCESS

Randomizing the Sample

Random sampling is used to collect a representative group of fish to accurately infer information about population size characteristics. Except for total weight, almost every statistic used to describe the sample should be able to accurately describe the population, e.g., mean, median, standard deviation, or variance of the weight, length, "height", etc. If a non-random sample is collected, inaccurate production information will be generated. As a result, unreliable predictions and comparisons will be made that will result in costly errors in production management. Crowding, net size, and sampling location are factors that should be considered to ensure that a random sample is obtained.

Net avoidance by larger or smaller fish is a common source of sample bias. Limiting the space in which the fish can move during sampling decreases the chance that fish that are more agile will move away from, or swim out of, the sampling net. Crowding can be stressful and crowded conditions should not be maintained for an extended period of time. During crowding, dissolved oxygen levels should be checked and adjusted. Dissolved oxygen levels should also be monitored if the fish that are removed for sampling are put into a temporary holding tank.

At an absolute minimum, the sampling net should be large enough to contain the largest fish in the population. A small net will bias the sample estimates towards smaller fish as the use of a smaller net increases the chance that a large fish will hit the lip of the net and escape instead of being captured. [Even a net equal to the largest fish is going to have this bias]. Patterns in the distribution of size within a tank can occur because fish develop preferences for tank regions; also, larger or stronger fish can displace smaller or weaker fish. This size assortment should be

considered when planning sampling location. If more than one net scoop of fish is needed to obtain an adequate sample size, fish should be taken from more than one location in the tank. If gentle crowding is used, a sample that represents the fish from all levels of the tank should be taken. Consider using a broad cast net in larger tanks. With some practice, you can become pretty efficient at doing this and it is a very good way to grab a sample without pre-alarming the fish. The first cast net thrown will collect the most unbiased sample.

The accuracy required determines the number of fish sampled; conversely, the sample size collected determines the accuracy. For a given accuracy requirement, the factor that influences sample size is the variance in the population. For example, in the case of 10,000 fish ranging in size between 100 g and 1,000 g requires a larger sample for the same accuracy as the same population of fish whose size ranges between 100 g and 150 g. As a rule of thumb for collecting weight data, at least 100 fish should be sampled. However, a minimum sample size of 30 will provide a rough estimate. If the total fish population is small – 200 fish or less – take a census to determine the characteristics of your population of fish without too much additional work. After collecting a sample, all of the fish collected should be measured in order to avoid sample bias. For example, suppose weight and length measurement are collected on 100 fish, but 23 remain in the holding tank. The remaining fish should also be measured to ensure a random sample. It is possible that the first 100 fish measured were the smaller, weaker fish that were the easiest to catch.

Weight measurements are used to: (1) estimate individual fish weight and growth rates over time, and (2) determine how much to feed. Because weight measurements are used for feeding, weight measurements need to be done at an early age. Weight measurements should be done from the time the fish are up and feeding, and producing feces.

A variety of techniques can be used for weight sampling. The choice depends upon the type of information needed and the time available. For weight data, scale sensitivity should be about 1% of fish weight. For example, if fish weight is 100 grams, the balance should display the weight to the nearest gram. If the balance is not sensitive enough to do this, consider bulk weighing multiple fish to improve accuracy. For example, if fish weight is 20 grams and the scale is accurate to the nearest gram, at least five fish at a time should be weighed.

Bulk weighing and counting results in the least amount of information, but is the least stressful for the fish, and takes the least amount of time. This technique may be used when only one person is available for sampling. By collecting bulk weight data, a measurement of total biomass can be obtained without measuring individual fish weights. Average fish weight can be estimated from bulk weight by counting the number of fish included in each sample of fish.

Procedure for obtaining a bulk weight

EQUIPMENT & SUPPLIES

- Scale – capacity great enough to weigh the mass of fish sampled
- Holding tank or section of the tank divided by netting (check for holes in nets!)
- Transfer Net
- Bucket – capacity large enough to hold the fish and water during weighing

PROCEDURE

1. Add sufficient water from the sample tank to bucket
2. Place the bucket on the scale
3. Tare the scale
4. Collect a random sample of about 100 fish and add the fish to the bucket
5. Record the total fish weight
6. Pour the fish into the holding tank and count them. Once a fish has been counted return it to the original tank
7. Calculate Average Fish Weight = $\dfrac{\textit{Total weight}}{\textit{Number of fish}}$
8. Repeat 3 times to get an average

Individual weighing produces more detailed data than bulk weighing. The data collected with this method can be used to calculate the variance or standard deviation, which are two statistics used for describing size variability in a tank. The additional information collected with this method can be used to decide when fish should be graded.

Example: Statistical Analysis of Fish Sampling

You have a tank of 5,000 fish. You estimate that the typical individual animal will weigh around 100 grams, based upon your last sample and a projected growth rate. You collect 3 samples of fish according to the above procedures, and the question is whether or not you have sufficient samples to estimate the population weight (average

fish weight of the tank). Justify either not taking another sample or that you should take an additional sample (see data in Table 15.9).

As can be seen in the example, Table 15.9, the first sample provided an unrealistic estimate of the average size fish in the tank. The average weight estimated in the tank changed by over 8% between the first and second samples. The third sample resulted in a less than 1% change in the estimate. As a general rule, keep taking samples until the estimate of fish weight changes by less than 2%.

Table 15.9 Statistical Example of Fish Sampling (weights in grams)

Sample Number	Sample 1	Sample 2	Sample 3
1	110	92	95
2	90	88	88
3	111	90	98
4	88	65	90
5	99	85	95
6	95	75	101
7	100	95	97
8	125	93	93
9	102	88	96
10	97	77	92
11	135	90	98
12	90	85	101
13	88	77	90
14	97	88	104
15	120	93	99
16	95	70	99
17	130	99	93
18	111	95	97
19	92	89	96
20	99	97	101
Average	103.7	86.6	96.2
St dev	14.2	9.2	4.2
St dev/average	13.7%	10.7%	4.4%
Mean of cumulative sample	103.7	95.1	95.5
Change in Mean	na	-8.3%	0.4%

Procedure for Obtaining Individual Weights

EQUIPMENT & SUPPLIES

- Scale – total capacity enough to weigh several fish
- Holding Tank or Net
- Transfer Net
- Bucket - sufficient to hold all the fish and water

Procedure

1. Add sufficient water from the sampled tank to the bucket
2. Place the bucket on the scale
3. Tare the Scale
4. Place a random sample of fish into the holding tank
5. Take one fish from the holding tank and place it in the bucket. Allow most of the water to drip from the net before putting the fish in the bucket
6. Record the weight and tare the scale
7. Repeat steps 5–6 until all the fish have been weighed
8. The bucket might begin to get full or fish could become stressed before all the fish have been weighed. If so, empty the bucket and put the fish back into the tank. Repeat steps 1 through 7 with fish from the holding tank until all the fish have been weighed.

Length information is used to:

- estimate growth rate in inches (cm), and
- when combined with weight, to estimate the condition factor (CF), which is used to track the body condition.

Length information is not used to calculate feeding rates. As a result, length measurements do not need to be done until fish are about two inches long. The CF is one of the most useful pieces of information obtained during the sampling process. With very few fish, the CF will tell you whether or not this particular cohort of fish is being under or over-fed, particularly when you have some historical data to go by. The CF will reach a fairly consistent value shortly after the fish have gone beyond the early fingerling stage. Use this CF value a lot! (See Chapter 3 for a listing of expected CF values by species).

Both weight and length can be measured very early in the fish growth phase to estimate feeding requirements and growth. 8- to 10 cm fingerlings (3 to 4 in) and larger fish will be much more resilient after sampling. However, at the sac fry stage and up to

about 2.5 cm (1 inch) the fish may need to be euthanized after taking length measurements due to the delicate condition of the young fish. Early feeding is usually based on a rough estimate, found by multiplying a typical value for sac fry weight times the number of sac fry present.

Length and Weight for Fry

Once the fish reach 5 cm (2 inches), length and weight measurements should be taken once every four weeks. While it is not necessary to sedate fry for weight measurements, they should be sedated for length measurements. Collecting length data is a two-person job—one person handles the fry; the other records data and monitors the fish during anesthesia and recovery. There are many standard definitions used for measuring length. One commonly used definition, the fork length, is measured from the tip of the snout to the base of fork in the tail. (Do not measure to the tip of the tail – fin erosion may shorten the tail and decrease measurement accuracy.) The equation to relate weight (Wt) to length (L) is (see Chapter 3 for more details):

$$Wt = \frac{CF(L)^3}{10^6} \tag{15.4}$$

EQUIPMENT & SUPPLIES

- Scale – total capacity enough to weight several fish
- Holding Tank or Net
- Transfer Net

After the equipment is set up, a random sample of about 50 fish is collected and placed into a container of water. Remove a few fish, place them into another smaller container, and slowly add the recommended amount of MS-222 solution (dissolved at a concentration of 1 gram per 100 mL of water). Add enough solution so that the fish are not swimming, but gill movement can still be seen.

Pick up the fish, blot with a paper towel, and place on dry towel or paper towel on top of the tarred scale. Record the weight and fork length (to the nearest mm) and place the fish into a bucket of fresh water for recovery from anesthesia. Length and weight should be matched for individual fish. (This matched data is used to calculate condition factors.) Repeat the process until all fish have been measured. Once all the fish are active in the recovery tank, move them to the rearing tank.

15.15 MISCELLANEOUS OPERATIONS

EUTHANASIA

The appropriate choice of a euthanasia method will differ depending upon whether the fish are being killed for diagnostic testing or are going to be processed for human consumption. In either case, the fish should be killed as humanely as possible and with pain and suffering minimized. For diagnostic purposes, using an overdose of an approved anesthetic such as MS-222 is a preferred and recommended approach. When anesthesia is used, a sufficient amount must be added to the water to stop the fish from breathing; usually a ten-minute exposure time is recommended.

When fish are to be used for human consumption, consult with a food science specialist to determine the most appropriate methods for your species as well as your market demands and constraints, e.g., if you intend to sell fish under some certification label, the certifying organization may have specific requirements on the method allowed to kill and process the fish. Do not chemically euthanize fish destined for human consumption. At this time no chemical is approved for this purpose. One method used commercially is to add carbon dioxide to the water column (works as an anesthetic) until swimming behavior has ceased, then proceed to the remaining necessary steps in the processing sequence, e.g., salmon processors will employ carbon dioxide, then use a head stunner followed by gill bleeding, and then move the salmon into an ice bath for cooling prior to processing or shipping. Cranial concussion (stunning) and direct cooling are not approved as humane forms of euthanasia nor is removing fish from the water and allowing them to suffocate. Producers are urged to keep abreast of the latest methods and forms of euthanasia for humane treatment of their fish.

ANESTHESIA

The purpose of anesthesia is to reduce fish stress for transport, sampling, or examination. Fish are generally anesthetized to a state of deep sedation, a condition that exists when the fish do not react to external stimuli except to very strong pressure, and exhibit slightly decreased opercular rate.

MS-222 is the only FDA-approved anesthetic for salmonids. FDA-approved dosage limitations are between 15 to 66 ppm for 6 to 48 hours for sedation, and a dose between 50 and 330 ppm for 1 to 40 minutes for deeper anesthesia. The dose is generally fine-tuned on the day of use. A 21-day withdrawal period must be observed prior to human consumption

of fish treated with MS-222. Further details on MS-222 usage can be found in the Appendix.

Two other options for anesthesia are carbon dioxide and sodium bicarbonate (baking soda). These drugs are not approved for treating food fish in the U.S. but are of "low regulatory priority". As an anesthetic, the recommended dose for CO_2 is 200 to 400 ppm for 4 minutes. For sodium bicarbonate, the recommended dose is 142-642 ppm for 5 minutes. (FDA has publically indicated that it considers usage of these materials below a specified level a "low regulatory priority." The doses recommended are below this level. Basically FDA has used this "low regulatory priority" language to permit the use of common aquaculture treatments with a long history of safe use to be used without going through the full regulatory approval process that would be required for each separate species.)

FISH HEALTH ANALYSIS (FHA)

Knowing how your fish behave when they are healthy is one of the keys to maintaining a healthy population of fish. This provides a basis of comparison when observing fish of questionable health. If the fish are sick, they will probably go through a period where they exhibit abnormal behavior in their swimming and feeding habits. By monitoring fish behavior, action can be taken before fish start dying, thereby averting large losses, or expensive chemical treatments. Check the dissolved oxygen, temperature, pH, feed condition, and the other easily monitored and/or controlled parameters before calling for a professional diagnosis. Do not make radical changes to system operation if unsure about the ramifications of the changes.

Sampling fish for a FHA can take various forms. Dead fish, sick fish, or random fish can be collected. These fish should be examined first in-house and, if possible, pictures taken for future reference. Cost increases as the complexity of the testing increases, so it is important to investigate the problem in-house if possible. Consult a fish health specialist for more information (see Chapter 16).

15.16 RECORD KEEPING AND MAINTENANCE

Maintain good records. Repeat ***Maintain Good Records***. Each cohort of fish that is grown should have a complete history, e.g., beginning and ending weights, types and quantities of feed fed, manufacturer used, general notes on water quality for the cohort in terms of temperature and oxygen.

Records should be kept of monitored water quality parameters as well as the general observations of the system manager. Recommended frequencies for monitoring water quality parameters are given in Table 15.10. A very simple daily record sheet can be created using a spreadsheet program that would show daily values for: day of month, feed consumed for day and cumulative feed used, water temperature, DO, TAN, NO_2-N, alkalinity, chlorine, a notes column, and a column for who recorded the data and the time. Spend effort in creating a data base management system so that you can visualize your results. Post your own standard growth curve and compare each cohort to your standard curve. Are they ahead or behind? Why?

Table 15.10 Schedule for Monitoring Water Quality in a Reuse System

Parameter	How measured	Frequency
Temperature	thermometer	daily
Dissolved Oxygen	meter or test kit	daily
Ammonia	test kit	twice per week[a]
Nitrite	test kit	twice per week[a]
pH	meter or test kit	twice per week
Alkalinity	test kit	weekly
Hardness	test kit	weekly
Chloride	test kit	weekly

[a]during startup periods, monitor daily (see Chapter 13)

The various mechanical components in an RAS should be checked frequently, with the frequency period being determined by how critical a component is, e.g., backup generator should be weekly, all pumps weekly, oxygen supply daily. Create a log of the maintenance schedule and have a column for completion of task, condition, action, and person responsible. One of the subtle dangers in RAS is that almost everything works all the time and the tendency is to become lazy in following maintenance protocols. Think of your RAS as a jet airliner that carries

several hundred people each flight. You as a passenger on this airplane probably appreciate the fact that a scheduled maintenance procedure is in place. Your fish are passengers in their airplane, the RAS. Check the systems regularly.

FISH RECORDS

Every aquaculture facility should have a husbandry plan. This plan lists the conditions under which the species will be raised. For a hatchery operation, details such as temperature for incubation and growout, feed formulation, number of eggs, and other special operational procedures should be listed in this plan. For a growout facility, details such as feed formulation, feeding rate, water quality requirements, expected growth rates, and other operational procedures. Incorporate any changes or improvements you make with each successive production cycle. This plan can be integrated with your predictions for capital expenditures as well. The purpose of the document is to (1) serve as a reference point during the course of the production (2) to serve as a reference document when comparing past production statistics. The details in this plan can help you locate inefficiencies in your operation that can be improved.

It is absolutely critical to keep complete records on fish production, feeding, water quality, fish leaving and entering the building. In addition, it is useful to keep records on system maintenance and visitors. Documentation is a key to gaining a thorough understanding of the system and achieving above average growth rates while minimizing costs. With good records, a producer can detect trends early on, before a problem can significantly affect growth rates or production costs. Such problems might include over feeding or decreased water flows. With this in mind, a series of suggested records are presented and discussed below.

DAILY FISH PRODUCTION SHEET

This data sheet is compiled to track fish production in each tank on a daily basis and the sheet should be set up for a month period. Information to be recorded on this sheet includes: stocking information, grading/sampling activities, fish movements among tanks, feed fed, disease treatments, and fish removed from the tank due to mortalities or sales. These sheets should be kept in a convenient place where the producer can refer to them often. It is best to keep them near each tank but this may not be feasible for some systems.

MONTHLY FISH PRODUCTION SUMMARY SHEET

On a monthly basis, after fish have been graded and moved, data from the daily fish production sheets should be transferred to the monthly fish production summary sheet so that it is accessible for easy reference. This summary sheet includes beginning and end-of-month totals for fish numbers and weights in tanks, total movements in and out of tanks, total feed fed, and overall feed conversions.

WATER QUALITY CHART

This data sheet is designed to record water temperature and oxygen levels in the oxygenation system and fish tanks. It is suggested that these records be made twice weekly. Both the air and water temperature should be taken about the same time of day. The dissolved oxygen level should be measured in the oxygenation system as water enters and leaves the tank. Water entering the oxygenation system will generally be 1 ppm below saturation (or about 8 to 10 ppm) and water leaving to the fish tanks should be supersaturated with oxygen to about 15 ppm or so. The producer is also provided space to record the dissolved oxygen level of the water leaving an average tank as well as that leaving a heavily loaded tank. These recordings will give the producer an overview of his system's performance. Space is also provided for comments and other notes such as when the water clouds up following heavy rains or when the weather suddenly changes as such events could trigger reduced feeding, etc. By noting such events, a producer would not be too alarmed if his fish went off feed for example. If the fish went off feed without such a "trigger event", then the observant producer would be on the lookout for possible water quality or disease problems.

EXPENSES

A data form is compiled to record all expenses during the year for the system. Sections should be provided for expenditures on fingerlings, feed, chemicals, electricity, oxygen, transportation, telephone, advertising, and miscellaneous costs.

INCOME FROM FISH SALES

Since this is the purpose of the operation, a form should be set up to help the producer keep track of annual fish sales.

PLANNING

Although the business plan has been written it is important to have yearly production plans which involve more detail than the business plan. The business plan is often a visionary statement, which may make forecasts for assumed real world conditions. While the initial production plan may have many of those visionary elements, as time progresses a good manager will be able to predict more accurately the timing of the production cycles as well as the profits and losses. Maximizing the difference between the expenditures and gross profit is an important goal in a production environment. Because prices are often dictated by market conditions, it is important for the manager to reduce expenditures, a variable within his or her control. Before starting a production plan, the manager should review the business plan to see what types of profits are expected and when, in addition to capital available for daily operations. As the operation progresses, the plan should be fine-tuned to increase the efficiency of the operation. Perhaps the most important component of your operation but perhaps the most overlooked.

Production planning allows you to find out where your costs are and what you need to do to turn a profit. If you can't figure out how to make a profit in the planning stage, you need to either consult for outside help or let you investors know. As difficult as it seems, it is better to lose $10 than win $100 and lose $1000. On the other hand, your plan may have you losing $100 and then making $1000. Either way, you as a manager need to have a plan of action while always realizing that it is subject to refinement. If you have co-workers or peers, share your plan, they may be able to suggest improvements or discuss dead-ends they have already encountered. As a manager, you need to be optimistic that you can make a profit but also be realistic if not.

IMPORTANT DATES

Regularly scheduled bill such as those for oxygen delivery, electric bill payments, feed orders, etc. need to be paid promptly to ensure service is not lost. Deadlines for permit applications should be tracked. As a planning tool, think about the fish's needs from birth (egg) to death. If automatically scheduled services are used, it remains the responsibility of the manager to ensure that payments are made in a timely manner

15.17 HOW TO COLLECT, ANALYZE & INTERPRET DATA

DAILY DATA COLLECTION

Use the "Appropriate Forms" to minimize the steps and the complications for getting data into the computer for analysis. The simplest way is to duplicate the format used by the spreadsheet, attach to a clip board, and place in critical locations throughout the facility. These forms should consider:

- Water Quality, Fish Behavior, Feed Consumption
- Accuracy of the data that is being collected

Try to keep data collection to a bare minimum, otherwise it becomes burdensome and is ignored. It is important to ask oneself a few questions before designing your data collection system:

- How does this help me to improve the status of my daily operations?
- Can I use this data in the future to establish trends, which may help me to more effectively predict or improve my production strategy?
- If this data indicates that I have a problem, can I do anything to correct the problem or is it out of my control?

DATA ANALYSIS PURPOSES

Record keeping is important not only for analysis after a harvest of fish, but is useful in monitoring fish growth and performance on an ongoing basis. Daily monitoring of data is important for all the following tasks:

- To forecast for production changes/improvements.
- To integrate the collection of data and later analysis.
- To learn how to see impending disasters and prepare for emergencies.
- Looking at both output and efficiency of production.
- Determining the maximum carrying capacity of the facility.
- Developing a 10-year production plan?
- Cost analysis and how to improve efficiency.
- Developing a yearly budget.
- Comparing current performance against production goals.
- Predicting expected mortality during the production cycle.

15.18 Care and Use of Laboratory Animals

For those of you who raise aquatic animals in a university setting or other government sponsored facility, you must be very aware of the Federal requirements on care and handling of animals. The Institute of Laboratory Animal Resources, Commission on Life Sciences, National Research Council, has published the "Guide for the Care and Use of Laboratory Animals" (National Academy Press, Washington DC 1996). This Guide emphasizes performance goals as opposed to engineering approaches. Using performance goals for the cultured fish places greater responsibility on the user and hopefully results in an enhancement to animal well-being. This is serious business. All farms and culture stations should obtain a Guide and review it thoroughly. Any Federally sponsored research must be conducted in accordance with the Guide.

15.19 References

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Chapter 16

Fish Health Management[1]

16.0 Biosecurity

For commercial success, an aquaculture operation must maintain fish at densities far greater than normally found in nature. The animals must survive and grow rapidly. Regardless of the culture system used, the fish producer must maintain an environment that supports good fish health.

Effective fish health management consists of practices and procedures that emphasize prevention of outbreaks of infectious and non-infectious disease. Implementation of biosecurity practices will reduce operating costs by minimizing the number and severity of infectious disease outbreaks. These practices and procedures should be documented in a facility "Fish Health Management Plan", which is available to the facility staff for periodic review and reference. An effective plan of disease outbreak prevention includes a monitoring protocol that detects fish health problems at an early stage. Running a facility without a prevention plan can be financially catastrophic, as it leads to continual responses to disease outbreaks as the fish health management strategy. This method of management results in added and unnecessary costs that include direct losses from mortality, inability to replace stock, facility

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closure orders, and restriction of movement orders. Market losses include reduced quality of survivors and a restricted market for healthy stock because of damage to a facility's reputation and missed markets. In addition to the direct costs incurred for diagnosis and treatment of an outbreak, additional costs are incurred in the form of diversion of management and labor from other essential tasks and underutilization of the production facility (Paterson et al. 1991).

Biosecurity consists of practices and procedures that:

- Reduce the risk that pathogens will be introduced to a facility
- Reduce tbe risk that pathogens will spread throughout the facility
- Reduce conditions that can increase susceptibility to infection and disease

Biosecurity cannot completely prevent entry of, or eliminate, all pathogens from a culture facility. Biosecurity accomplishes pathogen reduction rather than pathogen elimination.

Biosecurity is an important part of facility daily operating procedures. Planning should start during the design phase and protocols should be established before the facility comes on line. When biosecurity protocols are implemented in an existing operation, they may be perceived as being inconvenient because they were not considered when operational methods were developed or equipment was selected. Adding biosecurity as an afterthought may introduce an additional layer of complexity to an already inefficient operation. Effective biosecurity need not be an additional layer of work; procedures might be incorporated by modification of procedures already in use. Thinking about biosecurity before production begins allows non-intrusive routines to be developed rather than adding stopgap methods after problems arise.

Biosecure recirculating aquaculture system (RAS) husbandry requires that a system be designed so that it can be cleaned completely, easily and frequently. Any surface can serve as a substrate for microorganisms. All components of a recycle system including biofilters, low head oxygenators, CO_2 strippers, pipes and tanks should be constructed of nonporous materials and arranged to be easily accessible for cleaning and disinfection. Clean-outs should be installed to access in any part of the system for flushing of accumulated biosolids. Because wood cannot be easily and thoroughly disinfected, it should be considered only for fabrication of disposable temporary structures. Equipment and supplies should never be transferred from other locations to the facility.

16.1 Practices to Reduce the Risk of Pathogen Introduction

Water Supply

Entry of pathogens through a facility water supply is an important route of introduction, and will increase the risk for infectious disease outbreaks in aquaculture production systems. When possible, a groundwater supply should be used for the facility. Wells and springs do not usually contain resident fish, other aquatic animals, or aquatic invertebrates that could be pathogen carriers. If a pathogen-free water supply is at risk of contamination, or is unavailable, then influent water should be disinfected using ultraviolet radiation or ozonation. Well and spring water may need to be stripped of carbon dioxide and/or nitrogen gas, and oxygen may need to be added prior to using the water for fish culture. For small aquaculture operations of less than 100,000 lb per year (45,450 kg/yr), well water is the best choice because it will be specific-pathogen-free and constant water temperature and flow are more likely than with spring water. The well should be drilled and tested before the property is purchased. If the site does not have an adequate well water supply, another location should be selected. For larger operations, well water is also the best choice if an adequate supply is available. Surface waters harbor fish pathogens, and therefore, should be used only as a last resort, and then, only after effective sterilization (see Chapter 12). If spring water is used, it should be protected from animals that can carry fish pathogens, such as fish, birds, raccoons, salamanders, frogs and snakes.

Eggs and Fish

Entry of pathogens through the introduction of fish to a culture facility is another important risk factor for disease outbreaks in aquaculture. The risk that pathogens will enter a facility can be reduced by purchasing eggs and fish cultured in a disinfected or specific-pathogen-free (SPF) water supply and certified to be specific-pathogen-free. Certification involves testing a lot or stock of fish for specific fish pathogens relevant to the species and determining, based on statistical probability, whether they are free of those pathogens (Thoesen, 1994). Inspection is usually conducted once or twice per year. In general, in the case of egg purchase, the broodstock would be sampled and certified. In the case of fish purchase, a sub sample of fish would be examined for certification. Other options are maintenance of a pathogen-free brood

stock on site and/or use of quarantine (described at the end of this section) before fish are introduced to the production system.

> **"Rule of Thumb"**
>
> Every producer that transports fish directly from a pond into a recirculating system will experience catastrophic losses from infectious disease outbreaks.

Even if certified eggs are purchased, they should be disinfected upon arrival at the facility. The U.S. Food and Drug Administration (FDA) regulates the use of egg disinfectants in fish produced as food. Before using an egg disinfectant, verify that it is non-toxic for the intended species. Iodophors (organic iodine complexes with 1% active iodine) are generally used to disinfect salmonid eggs by creating a concentration in the water being treated of 100 mg iodophor/L water (100 ppm) for ten minutes after water hardening (Amend and Conte, 1982; Federal Joint Subcommittee on Aquaculture, 1994). As an example of how to treat 10 L of eggs and liquid, the following sample calculation is provided:

As purchased, 1% active iodine is 10,000 ppm (mg/L). Thus, 0.1 L (or 100 mL of Iodophor) will create a 100 mg/L ppm concentration in the 10 L of egg/liquid solution:

$$0.1\,L_{Iodophor} \cdot \frac{10{,}000\,mg_{Iodophor}}{L_{Iodophor}} = \frac{100\,mg_{Iodophor}}{L_{solution}} \cdot 10\,L_{solution}$$

One advantage of iodophors is the color change that occurs as it loses its effectiveness. Disinfection occurs while the amber color is present. Yellow or colorless iodophor is no longer effective. It should be noted that for some fish species, eggs are treated with either Fuller's earth (fine clay particles) or tannic acid to prevent aggregation of those eggs. Tannic acid is capable of inhibiting the disinfection qualities of iodophors (Cornwell, et al. in press). Unless there is extensive rinsing of the eggs following tannic acid treatment, the aquaculturist runs the risk of having in ineffective iodophor treatment of the eggs.

When purchasing eggs and fish, the number of different suppliers should be kept to a minimum to reduce the risk of pathogen introduction. Each supplier should be visited to determine whether the farm practices satisfactory biosecurity. If not, then fish should not be purchased from that source. One indication that the supplier is concerned about biosecurity is if a visitor is provided with pre-visit instructions, e.g., shower, clean clothes and clean footwear before arrival at the facility, no other facility visits the same day, and no contact with other aquatic environments.

FEED

Pathogens may be introduced into a recirculating system along with the fish feed. Commercial dry feeds are processed at high temperatures of about 160–180°F (71–82°C) for steam-pelleted, 180–200°F (82–93°C) for expanded and 220–350°F (104–177°C) for extruded feed, so pathogen introduction from this source is unlikely. However, as each bag (or lot) of feed is used, the lot number, and date manufactured and used, should be recorded in case trace back of feed needs to be carried out. Feed should be stored according to manufacturer's recommendations, usually stacked on a pallet with air circulation on all six sides. To avoid fish health problems related to rancidity or mycotoxins, feed should be used within the time recommended by the manufacturer.

Introduction of pathogens through live food presents a serious risk of contamination. All live food should be cultured in specific-pathogen-free conditions and should never be used from natural aquatic environments, e.g., ponds.

STAFF AND VISITORS

Pathogens can be carried into a facility by staff or visitors (human or animal). An employee and visitor parking area should be established on the periphery of the facility's grounds. Vehicles from an aquaculture facility, e.g., hauling trucks, should be disinfected upon arrival. The number of tours should be minimized and the number of people in a tour should be kept to a manageable size (three to six people; large groups should be broken into smaller groups). Visitors should be recorded in a logbook that includes name, affiliation, date, time and purpose of the visit.

Before they arrive at the facility, all new employees and all visitors should be given detailed instructions on compliance with biosecurity requirements. Personnel working in or visiting the facility should be required to change into clean coveralls and disinfected boots before entering the culture area. Visitors that are not from an aquaculture facility should be required to put coveralls over clothing, put on disinfected boots and wash hands with antibacterial soap before entering the production area. Visitors from an aquaculture facility should be required to remove their clothing, put on clean coveralls and boots and wash hands and arms for one minute. Visitors should be instructed not to

touch or lean against anything in the culture room. Barriers should be installed at the entry to areas that are off-limits, e.g., quarantine. After every tour, floors should be cleaned with disinfectant, e.g., 19 mL Roccal per gallon (5 mL/L) of water.

Employees should be discouraged from having aquatic pets at their homes and from working at another fish farm during non-work hours. They should not be allowed to bring pets into the fish culture area. Rodents, birds, other vertebrates and insects should be excluded from the fish culture area.

FOOTBATHS

Foot baths should be used at the entry to the production area (see Quarantine section for more detailed recommendations). They should be changed at least twice a week, more often if foot traffic is normally heavy. Foot baths should be changed after every tour and floors should be cleaned with disinfectant. An additional footbath can be placed "before" the primary footbath to serve as a pre-conditioning bath to remove surface soils prior to stepping into the primary bath. The pre-conditioning footbath can be changed frequently. See Fig. 16.1 showing Michael Timmons (one of the book authors) using the foot bath at the Freshwater Institute.

Figure 16.1 Entry area with foot bath prior to facility access.

QUARANTINE

Quarantine is the isolation of newly arrived fish. This isolation is imposed to prevent the spread of contagious disease to other fish in the facility. The quarantine facility should be designed for easy cleaning and disinfection. It should be a separate room or facility, not just a tank in the corner of the production facility. Waste discharge should be separate from the overall facilities systems and, if necessary, disinfected with

either ozone or ultraviolet radiation (see Chapter 11 Ozonation and UV-Irradiation).

Access to the quarantine facility should be restricted to a minimum number of people. No visitors should be allowed in the quarantine area. The area should be clearly marked with signs and directions as reminders to all company personnel and visitors. Quarantine equipment should be clearly marked and used only in the quarantine facility.

Work in the quarantine area should be saved as the last element of the work day. Personnel should wash hands and arms before going between the quarantine and production areas. Footwear should be disinfected. Clothing should be changed.

Traffic flow of personnel should be designed so that hand washing occurs as soon as personnel enter the area. Three sinks should be installed at the entry to the quarantine room. The first sink should be used for hand washing upon entry. Personnel should wash their hands with antibacterial soap, e.g., triclosan[2], for at least 30 seconds before they enter the quarantine area. After washing, hands should be dried with a clean paper towel and the faucet turned off with the paper towel before it is discarded.

The middle sink should contain disinfectant, e.g., 200 ppm chlorine, and be used to disinfect buckets, nets, meters and other equipment.[3] The third sink is used to rinse equipment after it is disinfected. (Regardless of the disinfectant used, it should be treated appropriately before discharge so that aquatic animals and plants are not harmed by the compound.) The doors and sinks should be located at a spot convenient to walk back and forth from tanks to the cleaning area.

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[2] Triclosan has been used as an antiseptic since the 1960s. It blocks an enzyme, known as "FabI," that bacteria need to manufacture the fatty acids used in cell membranes. Because animals possess a very different set of enzymes, triclosan does not interfere with this process in humans. This has led to its widespread use in over-the-counter preparations used on the skin or in the mouth. Once inside the cell, triclosan poisons a specific enzyme that many bacteria and funguses need for survival. Triclosan blocks the active site of an enzyme called enoyl-acyl carrier-protein reductase (ENR for short), preventing the bacteria from manufacturing fatty acids it needs for building cell membranes and other vital functions. Humans don't have this enzyme, so triclosan is harmless to them. One molecule of triclosan permanently disables an ENR molecule, which explains why triclosan has powerful antibiotic action even at very low concentrations. Triclocarban's structural similarity suggests a similar mode of action.

[3] One liter of 200 mg/L available chlorine is neutralized by 1.5 g of sodium thiosulfate.

Two disinfectant foot baths should be installed at the entry into quarantine. These foot baths should contain disinfectant, e.g., Roccal® at 10 mL per gallon of water, and should be filled at least 2 inches (5 cm) deep. The foot baths must be cleaned and changed as often as necessary to maintain efficacy; at minimum once per week.

Upon arrival, fish should be examined and all (not just a sample) of the shipment placed into quarantine. Fish should arrive in clean, debris-free shipping water and should be at least average in length and weight for their age, have normal skin color and no lesions on the skin or fins. Fish should be feeding and behaving normally within 24 hours after arrival.

An examination for parasites that includes wet mounts of skin scrapings and gill biopsies should be conducted the day of arrival. To determine which, and how many, bags should be sampled, the supplier should be asked if the fish were all collected from the same rearing unit. For each "lot" of fish, sample at least six fish with normal appearance and six fish with abnormal appearance. Throughout the period in quarantine, moribund fish should be examined for parasites and cultured for bacteria and viruses to determine whether pathogens are present that could threaten the remaining population of apparently healthy fish.

The quarantine period is often cited as thirty days. However, quarantine length for an individual facility could be greater or less than 30 days, depending on the species, age, source, and purpose of the fish. It should also account for incubation periods and development times for the pathogens that are known to present a risk, pathogen life cycles and expression of clinical disease in warmwater vs. coldwater conditions. Regardless of the quarantine period chosen, the addition of any fish to ongoing quarantine resets the clock to zero (Harms, 1993).

One objective of quarantine is to increase the probability that, if the fish are infected with pathogens, an outbreak will occur before fish are moved into the production system. Replication time for bacteria, viruses, protozoa and other pathogens is temperature-dependent. Some recommend maintaining fish at low densities to minimize stress; this practice will not create conditions that allow expression of disease agents that may be present. Consider keeping the water temperature within the optimum range for the targeted pathogen to speed up pathogen life cycles. Expose the fish to the same conditions, e.g., density, feeding, handling, they will encounter in the production systems, so that a problem may be detected before the fish are moved out of quarantine. A sub sample of fish can be stressed by exposing them for short periods to low DO concentrations, handling and/or disturbance such as bright lights

or motion outside tanks. These conditions will increase the likelihood that an infectious disease outbreak will occur.

Some protozoal pathogens, e.g., Ich, have a life cycle where some stages occur on and some occur off of (free-living), the fish. In these circumstances, fish can be transferred to a new tank in order to leave behind the free-living stage and reduce the number of parasites that are available to continue the infestation.

If an outbreak should occur while fish are in quarantine, they may or may not be moved into the production system. The decision whether to use these fish in production will depend on the pathogen involved, options for treatment and the ability to detect whether the pathogen is still present after treatment. For example, if a *Trichodina* sp. (ciliated protozoal pathogen) outbreak occurs, it can probably be eliminated by extension of the quarantine period and formaldehyde treatments followed by assessment of the remaining pathogen load through fish sampling and microscopic examination. However, if a viral disease outbreak occurs, there are usually no treatment options, elimination of the viral agent would be impossible and fish would not be moved to the production system.

Prophylactic antibiotics should NOT be used as part of a quarantine protocol. Prophylactic antibiotic use is illegal, will not eliminate pathogens from the fish, and can result in the development of bacteria resistant to the antibiotic.

About one to two weeks before fish are to be moved to the production system, environmental conditions should be gradually changed to mimic conditions in the production system. Alternatively, to allow fish to acclimate, production system water could be introduced before transfer to the production system.

16.2 Practices to Reduce Pathogen Spread

Meticulous Husbandry

Meticulous husbandry will reduce the risk that pathogens will spread throughout a facility. Foot baths should be installed at the entry to the culture areas (see Quarantine section) and should be cleaned and changed frequently. Personnel should wash their hands with anti-bacterial soap routinely and frequently. **Disinfectant** and rinse areas should be readily accessible for disinfecting buckets, nets, dissolved oxygen meters, thermometers, and other equipment. Cleaned equipment should be placed in a designated location in an equally clean area. For example, a disinfected net could be placed on a hook or a clean brush could be

placed into a disinfected trashcan for storage until the next use. Fish dip nets near tanks should be stored in a disinfectant rinse (see Fig. 16.2).

Decomposing debris provide a substrate for opportunistic pathogens to flourish. Therefore, tanks should be kept free of uneaten feed, feces, algae, and aquatic plants. Sumps, inflow and outflow pipes, aerators, spray bars, and any other equipment inside the tanks should be cleaned frequently. All parts of the system should be inspected, and cleaned if necessary, at least once per month.

Figure 16.2 Fish dip nets near tanks should be stored in a disinfectant rinse. Note: use of life preserver on model Margaret Timmons (daughter of one of the book authors), since tank depth is 3 m deep.

CULLING

To reduce the transmission of pathogens from fish to fish, dead and sick fish should be culled. At minimum, fish should be culled once a day. If possible, they should be removed on a continuous basis. Culled live fish should be humanely killed with an overdose of an anesthetic, e.g., tricaine methane sulfonate (MS-222) and not allowed to die from suffocation.

DISINFECTION OF TANKS AND EQUIPMENT

Empty tanks and equipment should be disinfected with 200 ppm chlorine for at least 1 hour. The chlorine should be neutralized with sodium thiosulfate, and rinsed before being used for a new group of fish.

> **"Rule of Thumb"**
>
> Chlorine, an effective disinfectant, **will kill fish** at even very low concentrations. Chlorine should be neutralized with sodium thiosulfate. One liter of 200 mg/L available chlorine is neutralized by 1.5 g of sodium thiosulfate.

Floors should be cleaned regularly. A clean pesticide sprayer may be used to spray the floors with disinfectant. The disinfectant should be allowed to remain for at least 15 minutes before rinsing off. When spraying disinfectant and rinsing the floor, the spray should be kept as low to the floor as possible to avoid propelling dirt and other debris into the air.

CULTURE ACTIVITIES

Culture activities should be scheduled to minimize the number of personnel working with a group of fish. If personnel resources are limited, unaffected tanks should be worked on before affected tanks. Younger fish should be cared for before older fish. Equipment that has touched the floor should not contact fish culture water. Any equipment that touches the floor or any contaminated surface should be disinfected before use in the culture system. Fish that have jumped from a tank onto the floor should not be returned to the tank. They should be humanely killed with an overdose of tricaine methanesulfonate (MS-222).

AEROSOL TRANSFER

Managers and designers of aquaculture systems, especially those with tanks aligned in close proximity should be aware that nets, boots, hands, fish, feed or water are not the only source of potential contamination. Aerosols can breach even well designed, isolated quarantine systems. Recent studies have shown that fish pathogens can be spread via the airborne route as aerosol/droplet sprays and can be detected in water downwind from an experimentally generated aerosol/droplet spray (Wooster and Bowser, 1996) and subsequently infect fish (Bishop, Smalls, Wooster and Bowser, 2003). This implies that fish pathogens can be spread via aerosol/droplet spray and could therefore move from tank to tank within an aquaculture facility, if a disease outbreak occurs in one tank. Solutions to help prevent the potential aerosol spread of pathogens include covers for aquaria or tanks, free standing or hanging wall barriers between tanks and changing

ventilation systems to limit spray or spread of aerosol plume. It has been reported in wastewater treatment plant literature (Brandi et al. 2000) that using diffusion aeration instead of mechanical agitation reduced dispersal of airborne pathogens from a wastewater treatment plant.

16.3 Reducing Susceptibility to Infection and Disease

Stress associated with handling, low water flow, poor nutrition, poor water quality and other husbandry characteristics will render fish more susceptible to, and aggravate the consequences of, infection with opportunistic and obligate pathogens. There are many strategies that can be used to increase fish vigor.

Optimum Nutrition

Poorly nourished fish are more susceptible to disease. The fish feed schedule and feed characteristics should be such that the fish receive the best nutrition possible.

Control Quality of Fish Stock

Eggs, fry and fingerlings should be purchased from optimum year class brood stock.

Reduction of Fish Stress

A wide variety of pathogens will infect cultured fish. Many disease agents are naturally present in low numbers in soil and water and do not cause problems because the fish are protected by natural defense mechanisms, i.e., undamaged skin, mucus, and cellular components of the immune system. However, when fish already crowded in culture operations are further stressed, their natural disease defense systems may be weakened, which reduces the ability of the fish to protect itself against infectious diseases. Catastrophic mortality that results from an infectious disease outbreak is often the result of, and response to, a stressful experience. Most infectious disease outbreaks can be avoided by proper management.

Culture and harvesting conditions and procedures should be designed to be as stress-free as possible. Poor water quality is a significant stressor that makes fish more vulnerable to disease outbreaks. Poorly nourished fish are also more susceptible. Light (excessive or rapid changes in intensity), noise and movement can stress fish and should be minimized. When netting fish, small rather than large net scoops should be taken. When grading, crowding should be done gently, without keeping too many fish together at one time. Fish must be handled during routine maintenance, stocking, and harvest. When fish are removed and processed, e.g., weighed, transported, they compensate physiologically, but there are limits. To reduce the trauma of handling, make sure all necessary materials, e.g., nets, hauling tanks, scales, and adequate personnel are immediately available. The use of salt (0.1–0.5% by weight in the water, see section 16.6 Treatment), aeration or oxygenation, and anesthetics (use according to FDA regulations) can reduce the stress associated with handling. Handle the fish gently and for as short a time as possible. If possible, do not handle fish that are already stressed or when culture conditions are marginal.

Vaccination

Vaccination should be used to help prevent infectious disease outbreaks. Vaccines are available in immersion or injectable form for many fish diseases.

16.4 Monitoring and Surveillance

By the time the major symptoms of poor fish health are noticeable, mortality has often begun and the problem is more difficult to solve. Therefore, the key to effective fish health management is a good program of monitoring, surveillance and response. Monitoring is an important part of early identification, isolation and treatment of a problem. If possible, this program should be established with the help of an aquaculture veterinarian.

How monitoring will be accomplished should be considered early on in facility design. Observation should be carried out daily. Fish condition and behavior can be observed through tank windows that are located to allow observation of areas where weak, sick fish normally congregate (at the inflow, in quieter areas of the tank). Culled fish should be periodically assayed for pathogens. Records on growth and feed conversion can be used to detect problems that are costing money, but haven't resulted in mortality.

Routine monitoring of water quality in a production system is imperative. When abnormal behavior is observed, water quality should be checked. If water quality is a problem, e.g., low dissolved oxygen, high unionized ammonia, corrective measures should be initiated quickly

to avoid more severe problems later. In an RAS, exchange of extra water can be very helpful ("...the solution to pollution is dilution..."). If abnormal behavior persists for more than two days, or death occurs, the farmer should seek professional assistance. In addition to a description of the history of the problem, water quality data should be provided to the veterinarian and/or diagnostic laboratory.

Even if fish recover quickly after correction of a water quality problem, the producer should think about the possible consequences days to weeks after the event. Behavior and feeding should be carefully monitored. Once or twice a week, a microscopic examination of gill tissue should be carried out to ensure that gills are recovering. Dying fish should be examined and should be cultured for pathogens.

Record Keeping

Record keeping is an important part of a fish health management program. Records should include number of dead and culled fish removed from culture tanks, observations of abnormalities, laboratory results and methods and results of treatment. This information can be used to improve biosecurity protocols and help a facility move toward an effective program of disease prevention and control.

Water Quality

The quality of the culture water has a major effect on fish health. Each species has an optimum range for water quality parameters. Some species are more tolerant than others when water quality deviates from this range. For example, tilapia are more tolerant than rainbow trout of reduced oxygen, high total suspended solids and elevated ammonia concentrations. In a properly managed recirculated system, dissolved oxygen and temperature remain relatively constant throughout the day and growing season, but alkalinity can drop to dangerously low levels during the nitrification process (see Chapter 7). In an improperly functioning or overstocked recirculated system, dramatic and rapid changes in dissolved oxygen, ammonia, or nitrite concentrations can result in high mortality of cultured fish or create stresses that can result in poor fish performance for weeks to months after the event.

Behavior

Abnormal behavior is often the first indication of an impending fish health problem. Personnel should become familiar with the normal behavior and appearance of the fish species in culture. All behaviors,

including feeding and swimming activity, and response to sudden movement outside the tank must be carefully observed. The fish producer must learn to distinguish nuances in behavior. For example, fish may normally move abruptly when the tank is approached. If they are sick they may not move at all or move abruptly and then become listless.

Healthy fish display "normal" behavior. For example, fish should feed vigorously when food is presented or shortly thereafter. In some culture tanks fish are usually invisible, except when feeding. In this environment, feeding behavior should be observed when automatic feeders are operating.

If feeding rates decline only slightly, fish may appear to be feeding normally. Such declines in feeding rate can be detected by closely monitoring growth rates and by checking for uneaten feed. Growth curves can be used to detect problems at an early stage.

Depending on the species, fish naturally swim in schools or alone. Distribution in tanks will vary by species, but is usually consistent within a species, e.g., some species prefer covered areas while others prefer uncovered areas; some concentrate toward the water inflow while others distribute themselves around the tank.

Behavioral and Physical Abnormalities

Table 16.1 lists abnormalities that may be observed when fish are sick. These signs will aid in the diagnosis of the cause of a problem. They should be monitored during daily fish observation, while removing dead or dying fish, and during sampling, grading, or carrying out any activity that creates an opportunity to examine fish closely.

Examination of fish gills is a useful, often underutilized, method for early detection of fish health problems. Gill tissue is a sensitive indicator of poor water quality conditions, i.e., elevated ammonia and nitrite concentrations, is susceptible to attack by parasitic and bacterial pathogens, and provides an entry point for systemic infection by pathogens. Fish producers should become familiar with normal gill appearance and carry out weekly microscopic examination of gill samples. Producers who have adopted this practice have found that gill examination is a worthwhile investment in time in return for the information it provides.

> **"Rule of Thumb"**
>
> Gill examination is a worthwhile investment in time in return for the information it provides. Know what a "normal" gill should look like!

Table 16.1 Fish Behavioral and Physical Signs for Stress and Sickness

Fish Behavior	Signs to Observe
Movement	Weak, erratic or lethargic swimming
	Increased or decreased reaction to external stimuli such as noise or movement
	Scratching, flashing, or rubbing against tank walls or bottom
	Twitching, darting, spinning or jumping out of the water
	Crowding near the influent water supply
	Swimming upside down
	Gasping at the water surface
Feeding	Not feeding
	Reduced feeding (detected by growth curves as well as observation)
Breathing	Decreased rate of opercular movement
	Increased rate of opercular movement
Physical Condition	Visible lesions or sores
	Cloudy eyes
	Protruding eyes
	Gills swollen, white, pink or pale red, eroded, puffy, bloody, brown
	Scale loss
	Swollen abdomen
	Excess mucous on the skin and/or gills (also, check for excess mucus on tank screens)
	Spots or fungus on skin
	Unusual colorations on body surface, including red swollen areas, gray or yellow lesions
	Flared opercula (gill covers)
	Frayed fins or tail
	Bubbles in eyes or in skin

16.5 Biosecurity Check

A consistently implemented biosecurity plan will make it easier to isolate the source of fish health problems. You must take precautions in a variety of ways to prevent the serious consequences of pathogen introduction into your facility. Once your facility has experienced infectious disease outbreaks, the costs associated with an effective cleanup are much larger than the costs incurred on a daily basis to reduce the risk of an outbreak. The following tables provide a guideline for producers working with or without consultation from a veterinarian on all aspects of biosecurity. The number of "yes" answers should be maximized for each of these sections, which have been adapted from Summerfelt et al. (2001).

Table 16.2 Personnel Management

Yes	No	**Personnel management**
____	____	Is frequent hand and arm washing with an antibacterial soap standard practice?
____	____	Are culture activities strategically scheduled to minimize the number of different personnel working with a particular group of fish?
____	____	Are vehicles disinfected before driving into the facility?
____	____	Has a visitor parking area been established at the periphery of the facility grounds?
____	____	Is facility access restricted to a minimum number of people?
____	____	Are the number of tours minimized and limited to small, easily managed groups of people?
____	____	Is a log book maintained?
____	____	Are visitors that have not been at an aquaculture facility within the past 48 hours required to put coveralls over clothing, put on disinfected boots and wash hands for at least 30 seconds with antibacterial soap?
____	____	Are visitors instructed not to touch anything in the culture room?
____	____	After every tour, are footbaths changed and the floor disinfected?

Table 16.3 Water and Fish

Yes	No	
____	____	Does the facility use a water supply that is free of fish, other aquatic animals, and aquatic invertebrates?
____	____	If fish and/or invertebrates are present, is the water supply disinfected using ozone or ultraviolet disinfection designed for target fish pathogens?
____	____	Is ozonation or ultraviolet radiation used to disinfect water within the recycle loop?
____	____	Does the facility design prevent the transmission of pathogens via air-borne water droplets?
____	____	Does the facility restock with eggs that are certified free of target pathogens or that were produced from broodfish that are certified free of target pathogens?
____	____	Are eggs disinfected on arrival at the facility?
____	____	If eggs are not used, is the facility restocked with fish that are certified free of target pathogens? Certified fry and fingerlings of some species are difficult to obtain if fry are reared in pond culture.
____	____	If eggs are unavailable and fish are purchased, are the fish quarantined when they arrive at the facility?
____	____	Are fish transferred to tanks in the facility without the introduction of water from the delivery truck or shipping bags?

Table 16.4 Fish Health

Yes	No	
____	____	Has a fish health monitoring plan been incorporated into the management of the facility?
____	____	Is water quality monitored and managed to maintain conditions appropriate for the species?
____	____	Is a premium quality feed fed to maintain optimal nutritional status?
____	____	Are fish handled gently to prevent injury?
____	____	Are eggs, fry and fingerlings purchased from optimum year class broodstock?
____	____	Is vaccination used as a disease prevention management tool?
____	____	Are abnormal, sick and dead fish culled to reduce the spread of pathogens?

Table 16.5 Quarantine

Yes	No	
____	____	Was a quarantine system planned early in the design of the facility?
____	____	Is the quarantine system in a separate building, room, or area where it is physically isolated from production?
____	____	Does quarantine use an independent, isolated culture system that uses a water supply and water discharge separate from the production system?
____	____	Is a flow-through water system or a system with a high proportion of make-up water used to dilute and flush pathogens from the system?
____	____	Does the duration of quarantine account for specific pathogen incubation and development times at the water temperatures for the rearing system?
____	____	When fish are added to ongoing quarantine is the clock reset to day zero?
____	____	Is the water temperature maintained at the upper end of the fish species optimum range to speed up pathogen life cycles?
____	____	On arrival, are new fish acclimated for 4 to 7 days to observe for abnormalities in appearance and behavior, and to confirm food consumption?
____	____	When abnormal fish are observed, are they sampled and examined?
____	____	If nothing abnormal is observed, are normal fish sampled and examined for target pathogens (parasites, bacteria, and viruses)?
____	____	As quarantine proceeds, are sub-samples of moribund and healthy fish taken periodically?
____	____	Are fish transferred to a new tank within the quarantine system if dealing with the possibility of stages of organisms that can be left behind when the fish are moved from the tank?
____	____	After the initial acclimation period, are fish held at culture densities they will encounter in the production system?
____	____	Do personnel wash hands and arms before going between the quarantine and production areas? Disinfect footware? Change clothing?
____	____	Is work in the quarantine area saved as the last element of the work day?
____	____	Is quarantine equipment used only in the quarantine area?
____	____	Are fish acclimated to production system water by introducing it to quarantine before transfer out of quarantine?

Table 16.6 Husbandry

Yes	No	
____	____	Are foot baths installed at the entry to culture areas and are they cleaned and changed frequently?
____	____	Do personnel wash hands and arms before entering the fish culture area?
____	____	Is a disinfectant and rinse area available for treating buckets, nets, dissolved oxygen meters, thermometers, and other equipment?
____	____	Is cleaned equipment stored in an equally clean area?
____	____	Are meticulous husbandry procedures followed for all life stages to remove feces, uneaten feed, algae, aquatic plants, dead fish, and decomposing debris from tank surfaces, inflow and outflow pipes, aerators, spray bars, and any other equipment inside the tanks?,
____	____	Are hands washed with antibacterial soap between working on separate cohorts of fish?
____	____	Is work scheduled so that the youngest, most vulnerable stages are tended first?
____	____	For tanks that are on the same recycle loop, is each tank treated as a discrete rearing unit and the potential for cross-contamination minimized?
____	____	Are tanks and equipment disinfected before use with a different group of fish?
____	____	Is the floor regarded as 'contaminated" and managed accordingly?
____	____	Are all parts of the system inspected and cleaned, if necessary, at least once per month?
____	____	Are floors cleaned on a regular basis?
____	____	Are pets, rodents, birds, other vertebrates, and insects excluded from the fish culture area?

16.6 Diagnosis

In the event of a fish health problem, an accurate diagnosis is essential. Each fish producer should locate an aquaculture veterinarian or fish health specialist who can make a visit to the farm when a fish health problem occurs. A correct diagnosis will determine the treatment regimen, including whether (and which) chemotherapeutics should be used. Attempts to diagnose and treat a problem based on hunches is usually not effective and misdiagnosis will result in wasted money and time, and further degradation in the condition of the fish. An incorrect diagnosis will also prevent the producer from developing an effective strategy to prevent a recurrence of the problem. If a veterinarian is unable to visit during a problem, then live fish may be shipped to a diagnostic laboratory, but the producer should be aware that without an onsite evaluation the recommendations will be unlikely to include a plan for prevention and control of future outbreaks.

Before shipping any fish off site for further diagnosis, the disease diagnostic laboratory should be called. Describe to the laboratory personnel the disease signs that you have observed and determine how they want you to prepare your fish for shipment. Do not expect a diagnosis over the telephone.

By informing the laboratory personnel of your problem and accurately answering their questions, you can facilitate a rapid and accurate diagnosis. The more information that you can provide to the diagnostic laboratory, the better will be the evaluation of your disease case. Some general guidelines in specimen collection, preparation, and shipment follow.

Specimen Quality

The quality of specimen submitted to a fish disease diagnostic laboratory will affect the quality of both the diagnosis and recommendations for corrective action. Dead fish are of little to no value for disease diagnosis because:

- Once death occurs, fish decompose very rapidly. If the disease is caused by a bacterium, other bacteria that grow during the normal decomposition process can quickly overgrow the pathogen and make identification difficult or impossible.

- Parasites require a live host for survival. When a fish dies, the parasites often quickly leave the fish in search of another live host.

- Viruses can be degraded by the compounds that are produced during decomposition. Once the fish dies the viruses may survive for only a limited period of time, sometimes only a few hours.

The veterinarian or fish health specialist at the diagnostic laboratory can provide guidance regarding the size and number of live fish to be shipped. The best way to transport sick fish to the diagnostic laboratory is for the producer to deliver them directly. Direct transport will provide optimum specimen quality, and will provide the fish health specialist with the opportunity to talk to the producer about the circumstances surrounding the mortalities. If requested, the producer should also bring a water sample from the culture system. This water sample should be collected in a clean container that can be capped tightly. If a chemical contaminant is suspected, the sample must be collected in a glass jar (not plastic) and handled according to instructions provided by the laboratory.

Packing and Shipping the Specimens

If fish must be shipped, the following methods (from most to least desirable) can be used:

- Live fish (most desirable)
- Iced fish
- Frozen fish
- Formalin fixed fish (least desirable)

Most diagnostic laboratories prefer specimens that are not preserved in formalin because the formalin will also fix (kill) pathogenic (disease-causing) microorganisms. Reaching a diagnosis may depend on the ability of the diagnostic laboratory to culture a bacterium or virus from the fish. Culture can be done only if the bacteria or viruses are still viable. In addition, a very important aspect of recommending a treatment for bacterial diseases is to determine the antibiotic susceptibility of the bacterium, which requires culture of the microorganism in the laboratory. Formalin-fixed materials can yield important diagnostic information following histological examination, but because of its limitations, most diagnostic laboratories prefer materials from which they can culture pathogenic microorganisms. This issue should be discussed with the diagnostic laboratory.

Fish shipped by any of the above methods should be collected from live fish in the system that are showing disease signs. It is best to collect the fish with a net or trap. Capture of fish by rod and reel will select

individual fish that are still feeding actively; the healthiest fish in the population. One of the first signs of many diseases is that fish stop feeding. Accurate diagnosis of the disease may not be possible as these fish may not yet be infected.

Live Fish

1. Obtain a strong, waterproof, insulated shipping container, e.g., a disposable Styrofoam cooler in a sturdy cardboard box, (see Fig. 16.3).

2. The veterinarian and diagnostic laboratory can provide advice about the ratio of fish to water volume. However, in order for sufficient oxygen exchange, the shipping bag should be no more than 1/3 full of clean water from the culture facility. Place the bag in the shipping container and add the fish. Fill the bag with pure oxygen or air. Seal the bag by twisting the open end tightly shut and securing it with several heavy-duty rubber bands or plastic tie-downs. An air tight seal is essential.

3. Water temperature during transport should be maintained within the species tolerance. For coldwater fish, place 3–5 pounds (1.5 to 3 kg) of crushed ice (or ice packs) in a strong plastic bag, seal the bag as described above, and place it in the shipping container next to the bagged fish. Warm water fish should not be packed with ice, and in cold weather hot packs may need to be included in box.

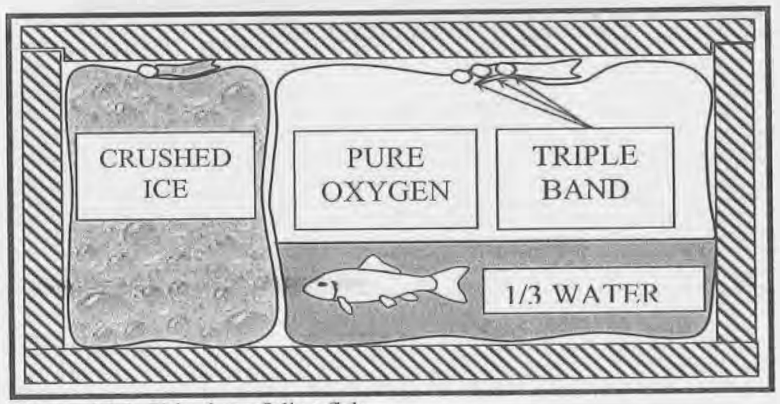

Figure 16.3 Shipping of live fish.

4. In a separate, small plastic bag place a note that includes name, address, telephone number, and information describing the fish and the culture system from which they came, e.g., why you suspect a disease; number of mortalities and their appearance; approximate size of diseased fish relative to other fish being cultured; when and how the shipped fish were collected; stock density; any known water quality parameters. Place the bagged note inside the shipping container.

5. Seal the shipping container. Be sure to indicate which end is "up" and that live fish are enclosed.

CAUTIONS: Take extra care to make sure the container won't leak. "Double bagging" can sometimes help. Ship via a carrier that can provide overnight delivery. It is always best to contact the fish diagnostic laboratory prior to any shipment and to coordinate the receipt of the fish with them.

ICED FISH

1. Obtain a strong, waterproof, insulated shipping container, e.g., a disposable Styrofoam cooler in a sturdy cardboard box, (see Fig. 16.4).

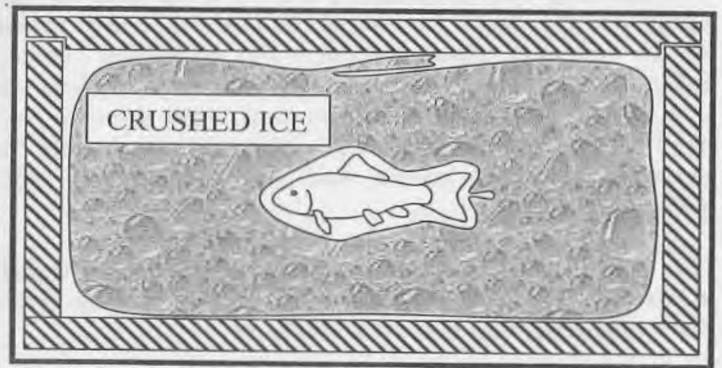

Figure 16.4 Fresh fish packed for shipment.

2. Wrap each fish individually with several sheets of newspaper to prevent freeze burns that may obscure signs of disease that could be diagnostically important. Place each fish in a separate plastic bag and seal the bag.

3. Place a larger, strong plastic bag in the shipping container and fill the bag with 2–4 inches (5 to 10 cm) of crushed ice.

4. Place the individually bagged fish on the crushed ice in the larger bag and cover them with an additional 2–4 inches (5 to 10 cm) of crushed ice. Seal the larger bag by twisting the open end shut and securing it with several heavy-duty rubber bands or plastic tie-downs. An air tight seal is essential.

5. In a separate, small bag place a note that includes your name, address, telephone number, and information describing the fish and the culture system from which they came, e.g., why you suspect a disease; number of mortalities and their appearance; approximate size of diseased fish relative to other fish being cultured; when and how the shipped fish were collected; stock density; any known water quality parameters. Place the bagged note inside the shipping container.

6. Seal the shipping container. Be sure to indicate which end is "up" and that iced (perishable) fish are enclosed.

CAUTIONS: Adequate amounts of crushed ice, usually 10–15 pounds (5 to 7 kg), will be satisfactory to keep the fish chilled during shipment. Ship via a carrier that can provide overnight delivery.

FROZEN FISH

1. Obtain a strong, waterproof, insulated shipping container, e.g., a disposable Styrofoam cooler in a sturdy cardboard box, (see Fig. 16.5).

2. Place each fish in an individual plastic bag and seal the bag. Freeze the fish in the individual plastic bags.

3. Place a larger, strong plastic bag in the shipping container and fill the bag with 2–4 inches (5 to 10 cm) of crushed ice.

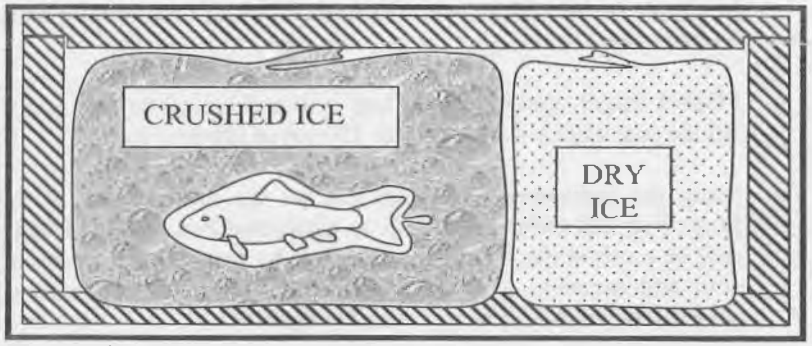

Figure 16.5 Frozen fish packed for shipment in Styrofoam box.

4. Place the individually bagged, frozen fish on the crushed ice. Cover the fish with additional crushed ice and tightly seal the bag by twisting the open end shut and securing it closed with strong rubber bands or a plastic tie-down. If possible, use dry ice to insure that the specimens do not thaw during transit. Five pounds of dry ice will normally keep specimens frozen for 24 to 36 hours, if the shipping container is well insulated.

5. In a separate, small bag place a note that includes your name, address, telephone number, and information describing the fish and the culture system from which they came, e.g., why you suspect a disease; number of mortalities and their appearance; approximate size of diseased fish relative to other fish being cultured; when and how the shipped fish were collected; stock density; any known water quality parameters. Place the bagged note inside the shipping container.

6. Seal the shipping container. Be sure to indicate which end is "up" and that frozen (perishable) fish are enclosed.

CAUTIONS: Check with the commercial carrier for their policy regarding shipment of packages containing dry ice. Ship via a carrier that can provide overnight delivery.

FORMALIN FIXED FISH

1. Make a 10% formalin solution. Neutral buffered formalin is best. Under practical field conditions, water from the culture facility will usually provide adequate buffering capacity to the solution. (To prepare the desired formalin solution, mix 9 parts water with 1 part formalin). Or, 10% neutral buffered formalin can be purchased and used directly from the bottle.

2. Kill the fish before placing them in the formalin solution. This can be done with an "overdose" of MS-222 (tricaine methane sulfonate at 1 gm per 500 mL H_2O). It is important that the fish be rapidly "fixed" by the formalin so that the quality of tissue preservation will yield useful information. Normally, formalin can rapidly fix tissues that are less than 1/2 inch (12 mm) thick. For this reason, the abdomen of larger fish must be opened for its entire length with one continuous cut. Most fish disease diagnostic laboratories will prefer to have the entire fish shipped rather than a limited number of tissues or organs. An "apparently normal organ" may yield valuable diagnostic information when examined microscopically. Questions regarding shipment of whole fish or selected tissues should be directed to the diagnostic laboratory before samples are sent. It is also important that adequate amounts of formalin be used to preserve the tissues. As a general rule, the **ratio of formalin solution to tissue must be 10:1** by weight or volume; i.e., 1000 mL of the formalin solution: 100 gm fish.

3. The container with the formalin and the tissue must be tightly sealed (see Fig. 16.6). Care should be taken to prevent breakage of the container. Glass containers can break and should be avoided. Use plastic bottles such as empty, clean food containers, e.g., peanut butter, mustard, salad dressing — use food service size for large fish, or containers obtained from scientific supply companies. Seal the container with tape to prevent leakage.

4. The sealed container should be placed in a shipping container filled with Styrofoam pellets or other suitable packing material. Care should be taken to prevent breakage.

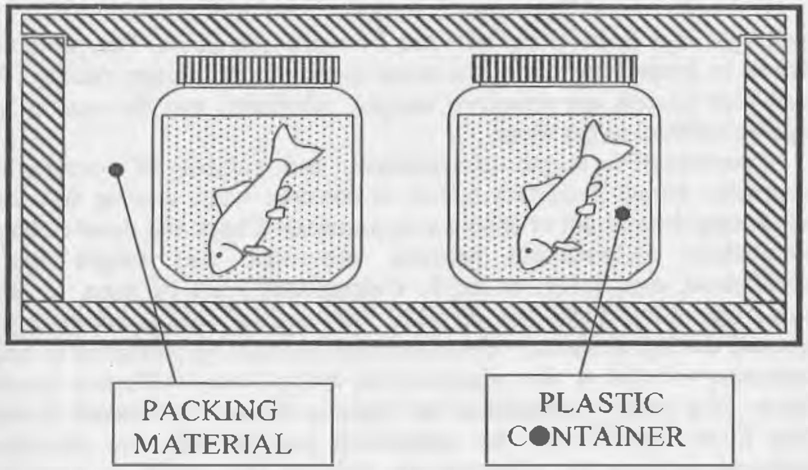

Figure 16.6 Formalin fixed fish packed for shipment.

5. In a separate, small bag place a note that includes your name, address, telephone number, and information describing the fish and the culture system from which they came, e.g., why you suspect a disease; number of mortalities and their appearance; approximate size of diseased fish relative to other fish being cultured; when and how the shipped fish were collected; stock density; any known water quality parameters. Place the bagged note inside the shipping container.

6. Seal the shipping container. Be sure to indicate which end is "up" and that preserved fish are enclosed.

CAUTIONS: Formalin is irritating and toxic. Provide and use good skin and eye protection as well as good room ventilation when using this chemical. A good practice when handling formalin or any other potentially irritating or toxic chemical is to use rubber gloves to protect the hands, goggles to protect the eyes, and a breathing mask to reduce inhalation of the chemical. An additional caution regarding formalin fixed tissues relates to current U.S. Department of Transportation regulations regarding the shipment of materials that are considered to be hazardous. These regulations cover shipments through commercial carriers. The regulations for proper packaging and labeling are very detailed and specific. Individuals who will ship formalin fixed specimens

must be properly trained by certified trainers before they can legally prepare packages for shipment. Unless an individual is capable of meeting these standards for shipment, it is recommended that they contact their diagnostic laboratory for alternatives.

Summary of Shipping Methods

The manner in which fish specimens are prepared and shipped will affect the quality of the information that can be obtained when the specimens are examined by the fish health specialist, Table 16.7. Working with a live fish will provide the diagnostician the best opportunity to gain useful information regarding the fish disease. The fish can be examined for live parasites. Microorganisms (bacteria and viruses) can be cultured from specimens that are delivered to the diagnostic laboratory alive and the sensitivity of bacterial pathogens to potential treatment chemicals can be determined. A histopathological examination can be performed on properly prepared organs and tissues. Tissue changes that are indicative of disease can be identified. It is critical that the tissues be preserved properly to insure that they represent the disease process and not decomposition after death.

Table 16.7 Impact of Fish Handling and Preservation Method on Disease Diagnosis

Shipment Method	Parasitology	Bacteriology	Virology	Histopathology
Live	+++	+++	+++	+++
Iced	+	++	+++	+/–
Frozen	–	++ / +	++ / +	–
Formalin Fixed	+ /–	–	–	+ +

Legend:

+++	no effect, excellent specimen for examination
++	negligible effect, good specimen for examination
+	moderate effect, specimen may be usable
+/–	substantial effect, specimen may not be useful
–	dramatic effect, specimen not useful

Specimens that are shipped on ice, or frozen, have some important diagnostic limitations. Living parasites may or may not be present on iced fish. When frozen fish are thawed, the shearing action of melting ice crystals will destroy parasitic protozoa, making their identification very difficult, if not impossible. The process of freezing and thawing tissues will also create a great deal of damage, making tissues of limited or no

use for histopathological examination. Iced and frozen specimens are normally satisfactory for the culture of bacteria and viruses.

Specimens shipped in formalin are useful for histopathological examination as long as the fish were collected live and were carefully preserved prior to shipment. As mentioned above, the diagnostician must be able to detect tissue changes that are caused by the disease process and not the result of decomposition after death. This can only be done with a carefully processed sample. The formalin fixed sample may not yield the identity of the disease organism and it will not provide information regarding the antibiotic sensitivity of a bacterium.

16.7 Treatment

Unless a veterinarian is involved with the investigation, the diagnostic laboratory will contact the farmer with the results of the laboratory tests. The disease will be identified and an appropriate and approved treatment or action will be recommended. In some cases, a change in management techniques will be necessary. In other cases a recommendation to add an antibiotic to the feed (for internal bacterial infections) or a chemical to the water (usually for external parasite infestations) will be made. It is extremely important that the producer follow the recommendations of the veterinarian or diagnostic laboratory and take appropriate precautions before a disease treatment is applied.[4]

One of the most important aspects of chemical treatment is knowing which chemicals are approved for use in food fish production. Each approved chemical can be effectively and legally used to treat a few to several diseases. No one chemical is appropriate for all diseases or situations. For instance, an antibiotic can be very effective in the treatment of a bacterial infection, but is useless if the disease is caused by a protozoan parasite. All chemicals have precautions and considerations associated with their use. If a farmer has no experience with a particular chemical, a small group of fish should be treated first, as a test before the entire lot is treated, to avoid potentially heavy losses due to chemical toxicity. Extreme caution should be practiced when applying any chemical treatment. Water quality influences the toxicity of certain

[4] Culling the sick fish is an alternative to treating with chemotherapeutics. When deciding whether to cull, treatment costs should be considered. Costs include the compound, labor during treatment and follow-up, risk of exposure to the other fish, additional losses of fish after treatment, possible decreased growth rates and the withdrawal time (waiting period) before fish can be sent to market.

chemicals and is adversely affected by some chemicals. The producer should be knowledgeable of the water quality in the culture facility. Of particular interest are dissolved oxygen, alkalinity, and the amount of organic material in the water.

Concentration, length of treatment, and number of consecutive treatments are all important factors to consider when treating fish and calculating the amount of chemical to purchase. Check and double-check calculations. Differentiate between volumetric and weight based calculations, e.g., mL/L or mg/L. Calculations must be done for the active ingredient. Flow rate or water volumes are critical numbers for accurate dosing. Accuracy of measurements should be confirmed to help determine whether or not treatments are being done at effective levels. Know drug safety information for humans (from the Material Safety Data Sheet (MSDS) for the compound) and for fish. For example, antibiotic overuse or skimping on dosages can result in bacterial resistance. Overdosing of antibiotics and sulfas can cause kidney damage in fish. Dilute the chemical before introduction into the tank, as this makes it safer to administer and helps distribute the chemical throughout the water more quickly to prevent overexposure of individual fish as the treatment is introduced into the water.

Fish should be taken off feed for 24 to 48 hours before treatment and, in most cases, should be kept off feed while it is carried out. This practice will reduce waste production and oxygen consumption during treatment. Fish should not be handled for 24 to 48 hours post-treatment.

In general, younger fish are more sensitive to chemical treatments than older fish. When there is any doubt, the treatment should be tested on a sub sample of fish before treating the whole population. Treatment should be discontinued and fresh water added if it results in thrashing, jumping, listlessness or any unexpected behavior.

Water quality will be affected by, and will affect, the treatment. For example, D.O. concentrations can decline during formaldehyde treatments. Temperature, pH, dissolved oxygen, alkalinity, organic loading and salinity can decrease efficacy or increase toxicity of a given treatment chemical.

Some chemical treatments can affect biofilter function by killing the nitrifying bacteria. System design should include plumbing that allows the biofilter to be isolated from the fish culture vessels. If the chemical concentration could be harmful to the biofilter, the tanks should be flushed after treatment with fresh water prior to reestablishing flow to the biofilter. However, it is possible that an untreated biofilter could become a reservoir for the disease organism. Effects of various treatment compounds on biological filtration are summarized in Table 16.8.

Table 16.8 Treatment Effects on Biofilter Function (Noble and Summerfelt, 1996)

Drug or Chemical	Treatment	Impaired function?
Benzalkonium chloride	2 ppm	yes
Chloramine-T	12 ppm	no
Chloramphenicol	50 ppm	no
Chlorotetracycline	10 ppm	yes
Copper sulfate	1.0 ppm	no
Copper sulfate	5 ppm	no
Copper sulfate	0.75 ppm for 24 hours followed by 0.5 ppm for 30 days	no
Copper sulfate	0.25 ppm for 24 hours twice at 3 day intervals, then a third one at 0.50 ppm, then one with 0.30–0.38 ppm continuously for 30 days	no
Erythromycin	50ppm	no
Formalin	low conc.	yes
Formalin	1:4000	no
Formalin	25 ppm in 3 indefinite treatments on alternate days	no
Formalin	15 ppm	low
Formalin	149 ppm indefinite	yes
Formalin	50-167 ppm for 1 hour with several treatments	no
Formalin	15–120 ppm indefinite with several treatments	no
Formalin	1:4000 for 1 hour	no
Formalin	35 ppm for 24 hours	yes
Formalin	153 ppm for 40 min, 3 times	no
Formalin and malachite green	25 ppm formalin with 0.10 ppm malachite green on alternate days	no
Furanace	0.1 ppm	low
Hyamine-3500	1.0 ppm	no
Hyamine-3500	2 ppm	yes
Hyamine-3500	1 ppm	yes
Hydrogen peroxide	100 ppm	yes
Malachite green	1.0 ppm	no
Malachite green	0.10 ppm on alternate days	no
Malachite green	0.5 ppm	low
Malachite green	3.0 ppm	no
Methylene blue	5 ppm	yes
Methylene blue	1.0 ppm	yes
Methylene blue	1 ppm	no
Nifupirinol	1 ppm	no
Oxytetracycline	Feed	no
Oxytetracycline	50 ppm	no
Potassium permanganate	4.0 ppm	no
Potassium permanganate	1 ppm	yes
Roccal™	0.067 ppm	no
Romet™	50 mg/day per kg fish for 5 days	no
Sodium chloride	3%	yes
Sodium chloride	0.5%	no
Sodium chloride	0.5%	no
Sodium chloride	1.5%	yes
Sulfadiazine	25 ppm	yes
Sulfamerazine	50 ppm	no
Sulfanilamide	25 ppm	yes
Terramycin	Feed	no
Trichlorfon	1 ppm	minimal

If the biofilter cannot be isolated during treatment and the chemical could harm the biofilter function, the chemical could be used at a lower concentration for a more prolonged treatment period. However, with this method a chemical may remain in the water longer than may be tolerated by the fish. An alternative is to treat the system at the therapeutic dose for a shorter period with the assumption that the chemical will remain in the water at an effective dosage as it is recirculated, but will be eliminated before adverse reactions appear in the fish.

Note that ***not all*** compounds listed in Table 16.8 are approved for use in fish. The aquaculturist should consult with their veterinarian or fish health specialist on compounds for which there is a current label (i.e., FDA approval) for use in fish.

Units of Measure for Chemical Application

The units of measure used in the following examples are primarily metric. Concentrations of chemicals are commonly expressed in terms of milligrams (mg) per liter (L) or parts per million (ppm). When making a chemical application in freshwater these two terms are functionally equivalent. One liter of water weighs 1 kilogram = 1,000 grams = 1,000,000 milligrams and 1 mg/1,000,000 mg (or 1 L) = 1 ppm.

Antibacterial compounds are added to the feed as a treatment for systemic (internal) bacterial infections. They are commonly referred to as rates. A generic expression is the weight of antibacterial compound per weight of fish per day for a specified number of days. This may be in terms of mg drug/kg fish weight/day. Historically, some antibacterial treatment rates have been expressed as a combination of English and metric units, e.g., g (gram) drug/lb fish weight/day. A list of conversion factors are provided in the Appendix to assist with calculations.

Methods

Methods for chemical treatment vary depending on the treatment compound, length and type of treatment, and the type of culture system. The supplier of the treatment chemical will provide instructions and precautions regarding its use. As a rule, to avoid adverse reactions between a chemical and metal, do not use metal tanks or metal measuring utensils when treating.

DIP

Dip treatments consist of dipping the fish or eggs into a chemical bath and then quickly removing them. Dips are generally used for treating external problems.

FLUSH

Flow through or flush treatment ensures that the water quality parameters such as ammonia and dissolved oxygen remain at acceptable levels during the duration of the treatment. This may be necessary for high density systems. The disadvantages of flush treatments are that (1) achieving adequate dilution for proper disposal may be a concern, and (2) the chemical costs can be high.

BATHS

Bath treatments are preferred to flush treatments, because they require less of the chemical(s), and are easier to manage. Since no fresh water is added during the treatment (definition of bath treatment), water quality will degrade and supplemental oxygen should be added. If the fish show adverse reactions to the treatment, then water must be exchanged quickly. Drain the treated water out of the system as rapidly as possible, then add fresh water. This procedure will remove virtually all of the treated water and should not overstress the fish.

CHEMICAL STORAGE

Always carefully read and follow the storage instructions for the chemicals you obtain. Never use any treatment chemicals beyond their expiration date. This is for both your safety and the safety of the fish. For example, formalin, when exposed to temperatures below 40°F (4.4°C), will form a white solid precipitate, paraformaldehyde, which is very toxic to fish. At this point, the formalin becomes unusable and must be discarded.

16.8 AQUACULTURE CHEMOTHERAPEUTICS

An aquaculture veterinarian will be able to provide information about regulatory issues surrounding the use of chemotherapeutics (drugs and chemicals used for treatment of disease). The fish producer must be aware of the legal consequences of chemotherapeutic use. A good practice is to maintain only those chemicals that do have specific

approval for aquaculture uses at the production facility. To an inspector, presence of non-approved chemicals at an aquaculture facility may imply use for fish treatment. Regulations concerning approved chemicals for use in aquaculture are continuously updated.

The use of these compounds in fish that are intended for human consumption is regulated by the US Food and Drug Administration. Before use, the producer should confirm that chemicals and drugs are approved for treatment of the target species at the intended dose and for the intended time interval.

Rules and regulations are described on the Center for Veterinary Medicine (FDA-CVM) website:

- http://www.fda.gov/cvm/index/aquaculture/aqualibtoc.htm

Veterinary biologics (vaccines, bacterins, diagnostics, etc, which are used to prevent or diagnose animal diseases) are regulated by the United States Department of Agriculture:

- http://www.aphis.usda.gov/vs/aqua/aquaphis.html

Environmental effects, i.e., discharge, of chemical treatments for food fish are regulated by the EPA (Environmental Protection Agency):

- http://es.epa.gov/oeca/ag/sectors/animals/anaqulaw.html#NPDES%20Permits

The FDA regulates chemotherapeutics at three status levels for drug approval: approved, not approved, and low-regulatory priority:

Level 1. Approved drugs have specific usage guidelines such as species, withdrawal time, and dosage and are only approved for specific manufacturers. Tricaine methanesulfonate (MS-222) and formalin are examples of approved drugs.

Level 2. Not approved or prohibited drugs may not be used in aquaculture. An example is malachite green.

Level 3. Low Regulatory Priority (LPR) drugs have specific applications under which they may be applied. There are five conditions that must be met when using these drugs:

- The substances are used for their listed indications.

- The substances are used at prescribed levels.
- The substances are used according to good management practices.
- The product is of an appropriate grade for use in food animals.
- There is not likely to be an adverse effect on the environment.

Note that any local laws may supersede national approval for drug usage. In addition, approval for use does not exempt the user from proper disposal, which must comply with national regulations such as NPDES (National Pollutant Discharge Elimination System) and state environmental regulations. Producers should become familiar with these regulations and formulate a compliant response before a fish health problem occurs.

Below are brief descriptions of some aquaculture chemotherapeutics. Approval status for each is listed with the compound along with precautions/considerations associated with their use.

AQUAFLOR

Aquaflor (florfenicol) is approved for treatment of enteric septicemia in channel catfish. The drug is only available for use under a Veterinary Feed Directive. This means that authorization for use of this drug can only be done with accomplished through a licensed veterinarian who has an established veterinarian-client-patient relationship. The drug is administered in the feed to provide a dose of 10 mg florfenicol (active ingredient) per kg fish weight per day for 10 days. A 12-day withdrawal period is required before fish can be slaughtered and used for human consumption.

COPPER SULFATE ($CuSO_4$)

Copper sulfate, an effective algaecide, is approved for use by the EPA. It can only be used for fish health problems if a producer has signed up to be part of an INAD[5] (investigational new animal drug) through the FDA. Copper sulfate is used to treat a variety of external parasites of fish, and can kill fish if used improperly. The relationship between toxicity of copper sulfate and alkalinity is very important. In water with an alkalinity less than 50 mg/L as $CaCO_3$, copper sulfate can

[5] An INAD allows a producer to use a drug or chemical while it is undergoing the review process for approval. INAD's are not available for all drugs. An aquaculture veterinarian can help a producer make the necessary contacts to become part of an INAD.

be very toxic to fish and should not be used unless a bioassay has been run in the water first with a limited number of the fish to be treated. The following general guidelines have been established for the use of copper sulfate in Table 16.9:

Since copper sulfate is an algaecide, consideration must be given to dissolved oxygen concentrations in the RAS. If the RAS already has low dissolved oxygen, an alternate treatment should be used.

FORMALIN-F, PARACIDE-F, AND PARASITE-S (FORMALIN)

Formalin is approved for use in the treatment of several external parasites. Formalin will remove 1 mg/L dissolved oxygen for every 5 mg/L of formalin used as a treatment. Therefore, if dissolved oxygen in a pond is low, aeration must be provided or a different treatment should be used. Formalin must be stored at temperatures above 40°F (4.4°C) because it will form very toxic paraformaldehyde at low temperatures. Caution should be used when storing this chemical at a hatchery with fluctuating air temperature.

Table 16.9 Permissible Treatment Concentration of Copper Sulfate as Affected by Alkalinity

Water Alkalinity (mg/L)	Permissible Treatment (mg/L)
0–49	Test for toxicity before use
50–99	0.5–0.75
100–149	0.75–1.00
150–200	1.00–2.00
>200	Ineffective; will precipitate As $CuCO_3$

POTASSIUM PERMANGANATE ($KMnO_4$)

Like copper sulfate, potassium permanganate ($KMnO_4$) is approved for use by the EPA, but not the FDA. The producer must be involved with an INAD through the FDA in order to use potassium permanganate. It has been used effectively against a number of external disease organisms of fish. The normal treatment is 2–8 mg/L, depending upon the amount of organic matter in the water to be treated. Ideally, a "wine red" water color should be maintained for a 12 hour period to ensure an effective treatment. A preliminary test can be performed with a small volume of culture water to determine the appropriate dose for the system.

Romet-30

Romet-30 is approved for the treatment of furunculosis in salmonids and enteric septicemia in channel catfish. It is used as a feed additive at a rate of 50 milligrams drug (active ingredient)/kilogram of fish weight/day for 5 days. A 42-day withdrawal period is required for salmonids and a 3-day withdrawal period is required for channel catfish before the fish may be slaughtered and used for human consumption. It is illegal for a producer to mix his/her own medicated feed.

Sodium Chloride (Salt or NaCl)

Sodium chloride (Salt or NaCl) is approved for aquaculture use as an "osmoregulatory enhancer". Salt can change the osmoregulatory balance (water balance) of aquatic organisms. It can control external parasitic protozoans by placing them in a condition of severe osmoregulatory shock. Care must be exercised to avoid over treatment, which will place the fish in the same condition of osmoregulatory shock. Sodium chloride is used as a 0.5% to 1.0% concentration in water as an indefinite (long term) treatment or as a 3% concentration in water for 10–30 minutes (stop the treatment earlier if the fish show signs of stress).

Sulfamerazine

Sulfamerazine is an antibiotic used at one time for the treatment of furunculosis in salmonid fishes. It was used as a feed additive at 10 grams of drug (active ingredient) per 100 pounds (45.4 kg) of fish weight/day for 14 days. A 21-day withdrawal period was required before fish could be slaughtered and used for human consumption. (Note: Old fish health literature implies that sulfamerazine is an approved compound for use on food fish, it is not! Because many individuals were substituting a generic "sulfa drug" for sulfamerazine, the manufacturer allowed its permit for this drug to lapse. Therefore, it is **not legal** to use this drug for food fish at this time.)

Terramycin

Terramycin is an antibiotic used to treat systemic (internal) bacterial infections. It is approved by the U. S. Food and Drug Administration (FDA) for the treatment of sensitive bacteria of the genera *Aeromonas, Pseudomonas,* and *Hemophilus* in salmonids and catfish. It is used as a feed additive at a rate of 2.5 grams of drug (active ingredient) per 100 pounds (45.4 kg) of fish weight/day for 10 days. A 21-day withdrawal period is required before the fish may be slaughtered and used for human consumption. It is illegal for a producer to mix his/her own medicated feed.

16.9 Treatment Calculations

Water treatments are based on water volume. A specified amount of chemical is added to a known quantity of water for a specified time. If too little chemical is added, the treatment will be ineffective; if too much is added or if the fish are left in contact with the chemical for too long, they may become stressed or die.

Feed treatments and fish injections are based on fish weight. A specified amount of drug is added to the feed (at the manufacturer) or injected into the fish. Incorrect dose may result in mortality or ineffective treatment.

Fish farmers should know the water volume of each culture unit, e.g., pond, tank, raceway, before a problem occurs, preferably recording this information when the system is designed or filled with water for the first time. The information should be stored so it is immediately available when needed. To become familiar with the procedure, the producer should practice making water volume calculations, (see Appendix). To obtain an effective, safe treatment, the ability to carry out accurate calculations is critical.

Sample Calculations

Three hypothetical situations are presented to provide the producer with an opportunity to become familiar with the methodology used to calculate fish disease treatments. For each example, there are several ways to correctly compute the amount of chemical to add or the drug to use. Calculations and steps are shown in detail for one method. These examples are intended to provide illustrate calculations only, not the entire treatment protocol. Chemotherapeutic dose and information about treatment duration or frequency and fish management before, during and after treatment should be obtained from a veterinarian.

Example 1

A tank system of rainbow trout is infected with the parasitic protozoan, *Ichthyophthirius*. The treatment to be used is copper sulfate ($CuSO_4$) and the producer is part of an INAD. The tank system contains 5,000 gallons of water with an alkalinity of 75 milligrams per liter (mg/L or ppm). How much $CuSO_4$ would you use?

Computation steps:

1) Examine Table 16.9 and determine what concentration of $CuSO_4$ should be added to the system to provide an appropriate and safe treatment.

You know that the alkalinity is 75 mg/L. Therefore, an appropriate treatment concentration for $CuSO_4$ is 0.5 mg/L.

2) Determine the quantity of $CuSO_4$ to be added to the tank to achieve the 0.5 mg/L concentration.

2a) *Convert the volume of the tank from gallons (gal) to liters (L).*

$$(5{,}000\,gal)\cdot\frac{3.8L}{gal}=19{,}000L$$

2b) *Determine a correction factor for the proportion of chemical ($CuSO_4$) that is active ingredient. $CuSO_4$ is 100% active.*

$$correction\ factor=\frac{1.00}{1.00\,(active\ ingredient)}=1.00$$

2c) *Compute the amount of chemical ($CuSO_4$) that should be added to the tank.*

$$(volume)\cdot(dosage\ CuSO_4)\cdot(correction\ factor)$$

$$19{,}000L\cdot\frac{0.5mg}{L\,CuSO_4}\cdot 1.00=9{,}500mg\ CuSO_4$$

2d) *Convert milligrams (mg) $CuSO_4$ to grams (g).*

$$9{,}500mg\cdot\frac{1.0\,g}{1{,}000mg}=9.5\,g\ CuSO_4\ \text{added to } 5{,}000\ gal\ tank$$

NOTE: *Ichthyophthirius* has a complicated life cycle, which must be considered in its treatment. Multiple treatments are required to effectively control outbreaks of *Ichthyophthirius*.

EXAMPLE 2

If alkalinity in Example 1 was below 50 mg/L, $CuSO_4$ would not have been the treatment of choice due to potential toxicity to the fish, 16.9. In this case, formalin would have been used as an alternative treatment.

Computation steps:

1) *Obtain a recommended treatment concentration for formalin from your diagnostic laboratory.*
An appropriate dose is 25 mg/L (or 25 ppm formalin)

2) *Determine the quantity of formalin to be added to the tank to achieve the 25 mg/L (25 ppm) concentration.*

2a) *Convert the volume of tank from gallons (gal) to liters (L).*

$$5{,}000\,gal\,\frac{3.8L}{gal}=19{,}000L$$

2b) *Determine a correction factor for the proportion of chemical (formalin) that is active ingredient.* NOTE: *Although formalin is 37% formaldehyde gas dissolved in water, for fish treatment purposes formalin is considered to be 100% active.*

$$correction\ factor=\frac{1.00}{1.00\,(active\ ingredient)}=1.00$$

2c) *Compute the amount of chemical (formalin) that should be added to the tank.*

(volume of tank) x (dosage of formalin) x (correction factor)

$$19{,}000L\,\frac{25mg\ formalin}{L}\,x\,1.00=475{,}000mg\ formalin$$

2d) *Since formalin is a liquid, it is desirable to convert milligrams (mg) to milliliters (mL).*

$$\frac{475{,}000mg\ formalin}{1{,}000mg\Big/1.0g} = 475g\ formalin$$

$$475g\ formalin \cdot \frac{1mL}{1g} = 475mL\ formalin\ (added\ to\ \text{tank})$$

EXAMPLE 3 FURUNCULOSIS

Rainbow trout in a recirculated system are infected with *Aeromonas salmonicida*, the bacteria associated with furunculosis outbreaks. The bacteria is sensitive to oxytetracycline. The fish will be treated by feeding Terramycin-medicated feed at 2.5 gm active ingredient/100 pounds of feed for 10 days. The fish are normally fed at 1% body weight per day and 10,000 pounds of fish need to be fed each day. How much Terramycin (TM) should be mixed with the daily feed ration in order to obtain the desired treatment?

Computation steps:

It is **illegal** for anyone but a feed manufacturer licensed by the FDA to make medicated feed. The producer should contact the feed manufacturer to determine what type of mix is normally available. If, for example, the feed is mixed at 2.5 gm active ingredient per pound of feed, then one lb of feed fed per 100 lbs fish will achieve the desired treatment. With 10,000 lbs fish normally fed at 1% body weight per day, 100 pounds of feed would be fed each day for 10 days. A total of 1,000 lbs of feed will be needed. Terramycin-medicated feed should not be fed at temperatures below 48.2°F (9°C). Fish cannot be marketed for 21 days post-treatment.

POST-TREATMENT REVIEW

When every new fish health problem occurs and when old problems keep recurring, a review should be done to determine the factors contributing to the problems – less than optimum environmental, e.g., water quality, conditions that lead to stress or directly to disease, ineffective biosecurity, and inaccurate measurement and poor management of culture conditions are common causes of recurring fish health problems. Good intuition can help isolate the problem; good record keeping can help to verify its origin so that strategies for early detection and prevention of the same problem in the future can be developed. Financial constraints, the design of the production system or constraints on the production cycle might preclude the development of strategies to prevent recurrence of the problem. However, strategies for early detection should be developed, so that even though the problem occurs again, early response can help mitigate the impact.

16.10 FISH DISEASE DIAGNOSTIC SERVICES

The importance of obtaining an accurate diagnosis for a disease problem cannot be overemphasized. The success of any treatment is absolutely tied to knowing the condition being treated and awareness of complicating factors. Accurate diagnosis of a fish disease requires specialized technical skills and appropriate laboratory facilities. Fish disease diagnostic services are available from a variety of sources in the United States. The fish farmer should establish a working relationship with an aquaculture veterinarian and/or contact a laboratory before a problem occurs to learn of any specific instructions associated with the submission of fish for diagnostic evaluation. For instance, some laboratories can, due to their funding, accept only submissions from restricted geographical locations; some fish health research laboratories accept only diagnostic cases that are referred by other diagnostic laboratories. Contacts for veterinary assistance and other diagnostic sources can be found at:

WWW.AQUAVETMED.INFO

or by contacting one of the USDA/CSREES Regional Aquaculture Centers (RAC's):

Northeastern Regional Aquaculture Center (NRAC)
http://www.nrac.umd.edu

North Central Regional Aquaculture Center (NCRAC)
http://www.ncrac.org
Southern Regional Aquaculture Center (SRAC)
http://www.msstate.edu/dept/srac/

Western Regional Aquaculture Center (WRAC)
www.fish.washington.edu/wrac/

The Center for Tropical and Subtropical Aquaculture
http://www.ctsa.org

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Chapter 17

Economic Realities & Management Issues

17.0 Introduction

This chapter is primarily based on the first author's personal involvement in a commercial startup Fingerlakes Aquaculture LLC (FLA) that began in 1996 in New York State. FLA was initially a 200,000 lb (91 tonne) per year, fully-integrated tilapia farm that included breeding stock, hatching, and growout operations. The farm was based upon the principles of recirculating aquaculture system (RAS) technology presented in this text. The farm later expanded to a production capacity of 1.2 million lb/yr (550 tonne). During this time, several rounds of financing occurred including capital injection by a venture capital firm. FLA's major market was the Chinese live markets, with a small amount of production targeted for the processed fillet markets. The first part of this chapter reviews some of the experiences associated with this commercial effort and the realities of indoor fish farming. For those who may be considering entering this business, read this chapter closely.

17.1 Case History of Fingerlakes Aquaculture (FLA) LLC

Four individuals created FLA in June of 1996: one of the authors (M.B. Timmons), his brother (marketing executive for a Fortune 500 company), Dr. William Youngs (Professor Emeritus Cornell University Department of Natural Resources), and a local businessman (owner of a lumber yard and apartment units). In addition, Drs. Dave Call (Dean Emeritus Cornell College of Agriculture and Life Science) and Gene German (Professor of Food Marketing, Cornell University) were members of the FLA Board of Managers. At that time, live market prices for tilapia sold at the farm, located in upstate New York, exceeded \$2.00/lb (\$4.40/kg). These market prices did not hold, however. Prices began to fall, and continued to deteriorate, and by late 1999, the market

price was just over $1.00/lb ($2.20/kg) at the farm. By 2002, the market price had recovered to a farm price of $1.40/lb ($3.08/kg) live weight, and for all of 2005 and 2006, the farm price has been at $2.00/lb ($4.40/kg) and sometimes higher. Predicting the price you will receive for your product is a real challenge. About all you can be sure of is that you will see 100% type swings in price and you better be prepared to handle cash flow subject to these large price variations.

In July 1998, FLA received a second major cash infusion from a successful entrepreneur. This individual had extensive and successful startup experience, which included agricultural and seafood types of businesses. Further rounds of financing were required in 1999 and 2000. Each new round of financing brought with it a significant dilution of the founding members' ownership percentage, but the personal guarantees and assignment of personal assets placed by these individuals to enable bank loans remained in place. These cash infusions and loans were simply part of a long struggle to increase production capacity from 200,000 lb/year (91,000 kg/yr) to the targeted 1.2 million lb (545,000 kg) per year. FLA did reach the projected production levels but it was a several year process to do this. FLA is now planning further expansion to a production volume that will make automated processing cost effective. FLA's success or at least longevity at being in business for such a long time can be attributed to several factors:

- Farm implemented in a series of stages (university prototype, small 100 tonne farm, followed by larger 500 tonne farm)
- Large scale operations
- Trained and motivated workforce
- Improved technology
- Adequate water
- Electrical rates below typical market prices ($0.04/kwh)
- Targeted marketing approach

A phased production program, i.e., a ramp up, is the most realistic approach to achieving the goals of your business. Most business plans start with some large scale operation, which is usually necessary to show a positive cash flow. Unfortunately, almost always, these operations, if funded, are unsuccessful. It is extremely difficult to achieve large scale success without having first built this capacity incrementally from smaller operations. This incremental increase in size and complexity of operations enables your management team to learn-by-doing wherein the inevitable mistakes will have a less devastating impact on a smaller operation than they would on a large scale operation. Given a sound technical approach, the ability of the management team to operate the business is the single most important factor in the determination of ultimate success or failure. Simply adding additional like-item systems of a proven and fixed design that is already working well is the best way to increase the size of the operation. If you choose to incorporate untested and unproven designs into your expansion plans, you will undoubtedly face unexpected problems that will compromise your ability to achieve the cost effectiveness that is predicted on paper (as you will be able to do with the programs presented in the Appendix). On the other hand, once you have a working system and management protocols refined at some reasonable scale, e.g., 200,000 lb (90 ton) per year, then this system or farm can be replicated as many times as necessary to reach the desired production levels and obtain the economies of scale necessary to achieve an economically competitive position.

"Rule of Thumb"

Adding additional identical systems of a proven design that is already working well is the best way to increase the size of your farm.

Finally, think a bit "out of the box". Being in aquaculture does not at all restrict you to producing a finfish food product that is going to generate only $1.00 to $3.00/lb ($0.45 to $1.40/kg). Why not consider aquarium fish, ornamentals, or coral? At least choose something you think you can enjoy working with.

17.2 Lessons Learned

Technology

The FLA team was very confident, perhaps even overconfident, that by using the full-scale systems developed, tested, and proven by Cornell R&D sites in this commercial implementation, there would be no major obstacles. This confidence was inspired by the knowledge that one of FLA principles had spent 15 years in leading a major R&D effort at Cornell University that was completely focused on indoor RAS technology. **This was a misplaced** confidence, because the Cornell system was based upon a different technology from what was initially implemented at FLA: microbead filters and continuous biomass loading at the university and fluidized sand filters at FLA. After a few months of working with high rate fluidized sand beds at the Cornell facility and being convinced that sand beds would ultimately prevail as the most

economically competitive biofilter system, FLA installed sand beds as the biofilter of choice (Note: FLA converted to microbead filters starting in 2002). The anticipated production level of the FLA's first facility was 400,000 lb (182,000 kg) per year but depended upon the Cornell system of continuous biomass loading, growout, and harvest. In this method, there is a mixture of size cohorts in a single tank with a harvest consisting of approximately 20% of the large fish being removed and then adding an equivalent number of small 75 gram fish back to the tank.

Even before FLA had placed their first fish into their systems (Fall 1997), they concluded that because of the lack of accurate, and timely data that could be generated from a continuous biomass loading, specifically the feed conversion and growth data associated with this method, FLA abandoned the continuous biomass loading approach in favor of a strict batch all-in-all-out loading. The batch loading method provides very accurate feed conversion and growth data at each tank harvest. FLA management felt that this data was the most critical information needed to define the overall economics of a large scale effort, which was the goal of FLA. The disadvantage of this switch from continuous loading to batch loading was that the batch process yields less production per year from the same sized facility.

FLA also made a change from the Cornell system of relying primarily on settling basins for solids removal to one that used mechanical screen filtration. No one at that time had any experience using mechanical screen filtration with the Cornell double drain design (see Chapter 5 Solids Capture) where the center drain concentrates waste by approximately 15 fold. We now know that micro screen filters must be re-sized (larger) if such a type of influent is to be used. Unfortunately, cost considerations frequently over-ride sound engineering judgment, particularly if there is no hard test or prototype results to verify the design. FLA erred by under-sizing their mechanical screen system, and this contributed to high suspended solids levels in the rearing tanks. This, in turn, compromised fish growth performance and overall production levels.

"Rule of Thumb"

Cost considerations frequently over-ride sound engineering judgment (this is a BAD thing)

FLA'S LARGEST ERROR—INSUFFICIENT WATER

It seems somewhat obvious that you should build a fish farm only where there is adequate water to support the expected operations. FLA's largest error was in not having an assured source of groundwater to operate the farm. Assuming that groundwater supplies would be adequate, FLA selected their initial site with other criteria in mind. These were important criteria, certainly, but not of the magnitude of the fundamental requirement for adequate water availability. **Never never never** commit to a site until you are convinced you have sufficient water to provide at least a 20% water volume exchange per day. Also, you should expect that the initial well recharge capacity will drop approximately 50% from its initial delivery levels (what your well driller demonstrates for one week of constant pumping during the exploratory phase), once the well is used on a continuous basis for a year. So, if you have 100,000 gallons (378 m^3) of standing water in your system, you need a well capacity of at least 20,000 gallons (75 m^3) per day. This means that your initial well capacities should be not less than 40,000 gallons per day (28 gpm, 106 Lpm).

"Rule of Thumb"

Before construction, identify/verify/establish a water source to replace 20% to 40% of system water volume on a daily basis (warm water) and 100% of system volume for a coldwater system.

To give you an idea of FLA's sad experience: FLA drilled five separate wells at the first farm site, four of them after the building was constructed when the first well that had initially provided 8 gpm (30 Lpm) of water during tests virtually dried up during construction. The primary author of this chapter believed, at that time, that FLA could successfully run the barn at planned production levels on about 8 to 10 gpm (30 to 38 Lpm) of continuous well flow. This proved to be not possible.

What typically is not accounted for in calculating water needs are:

1. *Loss of well capacity*
2. Purging requirements
3. Cleanup requirements
4. Flushing during tank emergencies
5. General usage around farm

FLA pursued a series of efforts to provide more water for the facility, including water recovery from the solids settling ponds with ground filtration recovery system followed by ozone treatment, but none of these attempted solutions were satisfactory. You must have water, in at least the quantities described above. Eventually, FLA abandoned the site at considerable expense, and moved to a new location that had access to municipal water supplies with the capability to deliver several hundred gallons per minute more than the nominal needs of the facility.

MANAGEMENT

Management is the most critical component of any aquaculture venture. From its inception, FLA had an extremely strong management team in place. The General Manager was recruited from his position as a GM of a large (1 million lb/yr or 455,000 kg/yr) integrated salmon farm. Dr. Youngs had started, successfully operated, and eventually sold a large trout farm. The board members were a good mix of business leaders and marketing experts. Even with this experienced management team, FLA barely survived. If FLA had tried a larger scale of effort than their initial farm that consisted of breeding, fingerling, and target growout of 250,000 lb (114,000 kg) per year, we predict they would have failed categorically. FLA produced 168,000 lb (76,000 kg) of sales in their first year of production compared to their target of 250,000 lbs (114,000 kg).

> **"Rule of Thumb"**
>
> Management is the most critical component of any aquaculture venture.

FINANCING

Regardless of all you hear about government guaranteed loans for business startups, in the FLA experience, it came down to equity infusion (self and outside investors) and personal guarantees of loans. The banks will take no risk with their depositors' money. They assume you will fail. They also assume that the value of your assets will be practically negligible. In truth, these are probably reasonable assumptions, particularly regarding the residual value of assets and equipment. The market value for used fish equipment is probably 10 cents on the dollar at best. Yes, you can probably sell common industrial equipment, such as your forklift for a reasonable salvage, but more specialized equipment, such as pumps and other fish equipment are of little to no value.

Therefore, the bank will require that any loans to the company be personally guaranteed and may in fact require these loans to be perfected. This means that the guarantors must place assets equal to or exceeding the value of the loans in the bank's control. Then, if necessary, the bank can liquidate these assets if the terms of the loan are not being met.

> **"Rule of Thumb"**
>
> Assume 10 cents on the dollar for used fish equipment.

The process of finding adequate financing requires considerable time and effort. But be prepared for the realization that generally all mechanisms of financing and leasing will end up at the same point, wherein the loan/lease will require equity guarantees. Also, bear in mind that these activities require tremendous time and effort by someone to pursue potential sources, develop detailed presentation materials, and attending meeting after meeting. Assume a successful loan closing will take at least three months before any money is actually available for you to use, and this is after you have found a bank that has said they will loan you the money!

Venture capital (VC) firms are also supposed sources of capital. FLA's experience with these during the initial rounds of seeking capital was that the VC assumed that what you had was probably not unique; that if you could do it, so could countless others; or that the overall potential to make money in a food related business was not attractive, especially in comparison to other investment alternatives available to them. So, unless, you have an inside track with a particular venture firm, the likelihood of obtaining equity from them is not high—and even when you do have personal contacts, it is still not a likely outcome.

On the other hand, if you can demonstrate some Phase I success and bring investors to see your operating farm that has demonstrated your capability to breed and produce fingerlings successfully, grow fish with reasonable performance values, and sell fish successfully, then you become much more likely to generate VC or other outside investor funding. The key of course is to achieve success at the first level of an operating farm.

17.3 INVESTMENT CHOICES

In our roles as university researchers, extension professors, and consultants, we are often involved in helping people to decide if entering aquaculture is a wise investment decision. In particular, should an

investment be made? Our opinion is that very few groups or individuals are capable of raising fish, using RAS technology or any other method. We would not recommend anyone invest in someone else's aquaculture operation until that group had successfully demonstrated their capability to raise fish. Then, based upon their initial results, assess the likelihood of the company being profitable in an expansion phase. In this assessment, be sure to use realistic values for product selling prices to predict business profits. We have often seen business plans that use retail prices where the farm's only realistic opportunity to sell product will be in the wholesale market or even worse to a hauler/broker who buys your fish and sells into the wholesale markets. Some business plans will even confuse fillet pricing with in-the-round pricing (whole fish, non-gutted). Be careful and be realistic.

17.4 Species Selection

A general theme for those entering aquaculture is to choose a high value species, such as perch, walleye, or ornamental fish. The primary reason these fish command high market values is that their supply is very limited. As aquaculture develops successfully for a particular species and the market supply dramatically increases, the market price dramatically reduces. Also, do not be misled by these scenarios of apparently lucrative markets. There are reasons that the market is under supplied and driving the high price. The salmon and striped bass industries experienced market price reductions of roughly 50% in less than a year's time during their history. Tilapia prices in the US seem to go through about a 5 year cycle and will cycle from \$1.00/lb (\$2.20/kg) to \$2.00/lb (\$4.40/kg). When farms are being paid prices at the top of the cycle, farmers make good profits. This brings more production (new producers) on line and this then drives the price down, since the demand for live tilapia has remained constant for the last 20 years of between 16 to 20 million (whole fish basis) lbs per year (7,000 to 9,000 tons). Meanwhile over this same time frame, the market supply for fillets has increased from essentially nothing to 250,000 tons (for 2006)!

To best predict price trends, one should compare current market prices for a particular species to some other commodity type fish, such as farmed salmon or catfish. These fish provide alternative choices to the consumer, thus bracketing the range of pricing that a consumer is willing to pay. Other premium meats also provide a substitute for the meat purchaser. For example, the catfish industry produces 600 million lb (273,000 ton) per year in comparison to the US yearly production of

tilapia of 18 million lb (8,200 ton). Pond side prices paid to the farmer for catfish are generally around \$0.75/lb (\$1.65/kg) on a whole-fish basis. Fillet yields for catfish are 45% and tilapia yields are now at best around 33%. Thus, for tilapia fillets to be competitive with catfish fillets, the equivalent farm price for tilapia would have to be \$0.55/lb (\$1.21/kg), which may indicate that the current farm prices of \$1.00/lb (\$2.20/kg) could fall even more. Genetic improvements in tilapia carcass yield will balance the pricing, but until then, large scale tilapia farming will have to be competitive with catfish farm prices as noted above. Similar pricing logic should be applied to other species. Remember, farm tank-side price can drop by half within 6 months. It has happened in the past, and it will happen again. Be forewarned and prepared.

"Rule of Thumb"

Success in culturing high-value species will result in dramatic drops in their market price.

17.5 Competitiveness of RAS

If RAS technology is going to be successful, then the aquaculturalist must expect to compete against the other commodity meats and large scale fish farming such as currently being practiced by the net-pen salmon industry or the US catfish industry. (See Figure 17.1 for historical production levels).

The dismal truth is that there has been 20 or more years of generally negative results and viability associated with RAS. Generalizing, most of the problems have not been so much related to the technology as the miss-management of systems and attempts at growing species that were not suited towards RAS. The authors also disagree with the idea that RAS can only be used to produce high value products. Like any business, success is built upon a whole series of critical factors; any one of which that is missing will lead to a business failure.

General miss-perceptions that are commonly associated with RAS technology include:

- Overly complicated,
- Prone to catastrophic failure
- Expensive,
- Suited only for high-value species
- Only highly educated people can be trained in their use

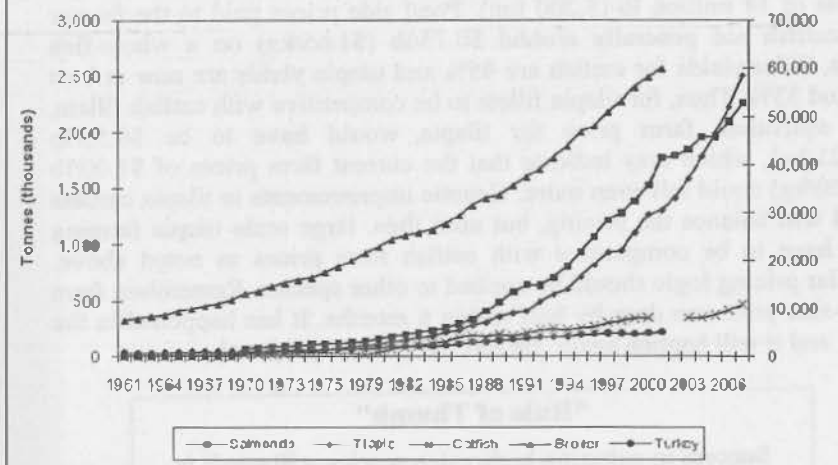

Figure 17.1 World yearly production of salmonids, tilapia and catfish (FAO, 2007).

These labels may have been warranted a few years ago, but today, RAS technology is none of the above. Of course, you can make any technology overly complicated, expensive, and prone to failure-- but we are well beyond that (or at least we should be) in the RAS industry today. The unit processes associated with a RAS have been previously discussed in earlier chapters. While all unit processes available will not typically be used in any particular RAS application, all these processes should be considered during the design and planning stages, particularly in relation to the level of water quality control and quality desired. The challenge is to design a system that is matched against the water quality requirements of the targeted species and to do this in a cost-effective manner.

17.6 INFRASTRUCTURE & CAPITALIZATION

An aquaculture development project will require significant infrastructure in terms of water, waste disposal capacity, enclosed building space, electrical energy and load demand supply, and transportation logistics. While each site considered will require a thorough engineering analysis, approximate minimal site requirements are given in Table 17.1.

Table 17.1 Approximate Infrastructure and Utility Requirements

	Cool Water (Trout)	Warm Water (Tilapia)
Production per year (kg)	454,000	454,000
Footprint of buildings, m^2	5,600	3,700
Water Required & Discharge per day, m^3	3,000	300
Heating Requirements, MJ/day (peak seasonal demand)	20,000	40,000
Electrical Requirements, kWh/day	6,000	4,000
Liquid Oxygen, m^3/day	1,000	1,000

WATER SOURCE

The *major advantage* of RAS is that the water requirements for production are reduced dramatically (see Chapter 1 Introduction to RAS). What new water is introduced into a RAS must be biologically secure. The major vector for introducing disease organisms into a production site is through the water or through the animals being brought into the farm. If both of these vectors are clean, then the occurrence for losses due to disease is practically non existent.

Great effort must go into making a farm biologically secure. The source water ideally should be deep wells providing drinking water quality water. There is no substitute for a biologically secure source of water. Do not build on any site until water sourcing issues are established. You should assume that as a minimum site requirement, you will need one system volume of water per day for cold water systems and 20-40% for warm water systems, even though typical usage rates will be 20% of system volume per day or less. Treating non-biosecure water sources will require some combination of mechanical filtering, ozone, ultraviolet, and chemical processes. (See Chapter 5 Solids Capture, Chapter 11 Ozonation & UV-Irradiation, and Chapter 16 Fish Health Management).

The quality of the water necessary will depend upon the species being grown and the stage of production being implemented. Water quality for an egg rearing operation will be more stringent than an advanced growout system for tilapia. In terms of water quality, criteria must be specific to species and stage of production.

CAPITALIZATION COSTS AND UNIT PROCESSES NEEDED

A list of the most important unit processes of indoor recirculating aquaculture and the water quality parameters they address are presented below:

Table 17.2 Unit Processes Needed for Water Quality Control

Unit Process	Water Quality Parameter Addressed
Biofiltration	Ammonia and Nitrite Nitrogen Removal
Solids Separation	Excess feed and fish waste removal
Carbon Dioxide Stripping	Carbon dioxide concentration in water
Oxygenation	Dissolved oxygen concentration
pH Balance	pH, CO_2 concentration, Alkalinity

The equipment used to perform these individual unit processes all contribute to overall capitalization costs. Economically competitive food fish production will depend upon collectively reducing capitalization costs to be at least nearly as efficient as the salmon industry, e.g. $0.40/kg per year of system capacity production (see calculation later in this section). Inventive new ideas or management methods must be developed as to how to combine unit operations or to reduce costs associated with present technologies. Probably more than any other factor that can contribute towards this goal is to increase the scale of the production operations. Just as dairy, hogs and poultry have increased production per farm and therein improved labor efficiency and other cost of goods components, the aquaculture RAS based industry must also do so.

A photo representative of a current intensive RAS used for raising artic charr is shown in Figure 17.2 .

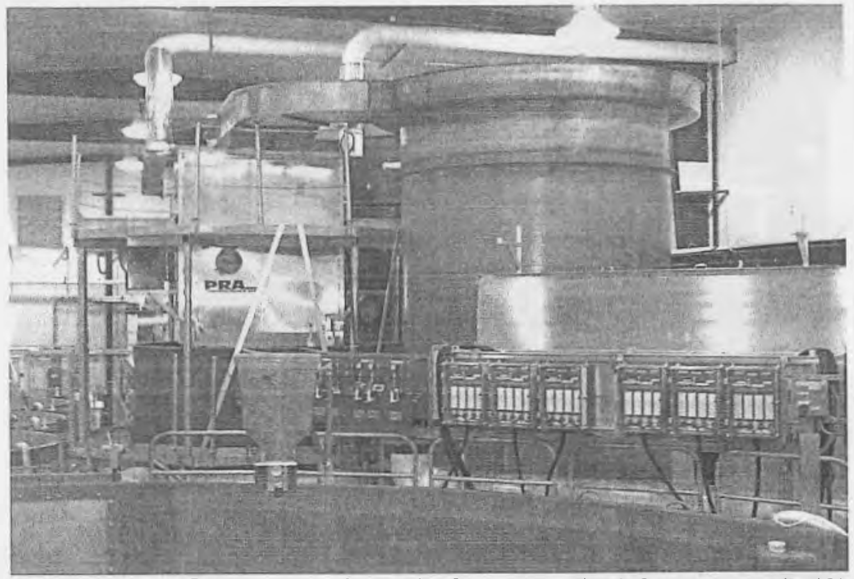

Figure 17.2 A RAS using a CycloBio filter, Low Head Oxygenation (LHO) unit and stripping columns. Water flow exiting the top of the fluidized-sand biofilter flows by gravity through a cascade stripping column, an LHO unit, and a UV irradiation unit before being piped by gravity to the culture tank. Photo courtesy of the Conservation Fund Freshwater Institute (CFFI, Shepherdstown, WV).

The Fingerlakes Aquaculture system (Groton, NY) that is used for rearing of tilapia is shown in Figure 17.3 and a simplified unit process diagram for this system is shown in Figure 17.4 (from Fingerlakes Aquaculture, Groton, NY). Note the differences in complexity based upon the use of the CFFI system being used to rear artic char (a sensitive water quality species) versus the Fingerlakes system that is used to raise tilapia (a less sensitive animal to water quality conditions).

Figure 17.3 Overview of a CycloBio System at Fingerlakes Aquaculture (10 meter diameter fish tank is show in front of CycloBio; note mixing fans that are blowing air through water fall from top of distribution channel leaving CycloBio and delivering water to LHO units). Photo courtesy of Fingerlakes Aquaculture (Groton, NY).

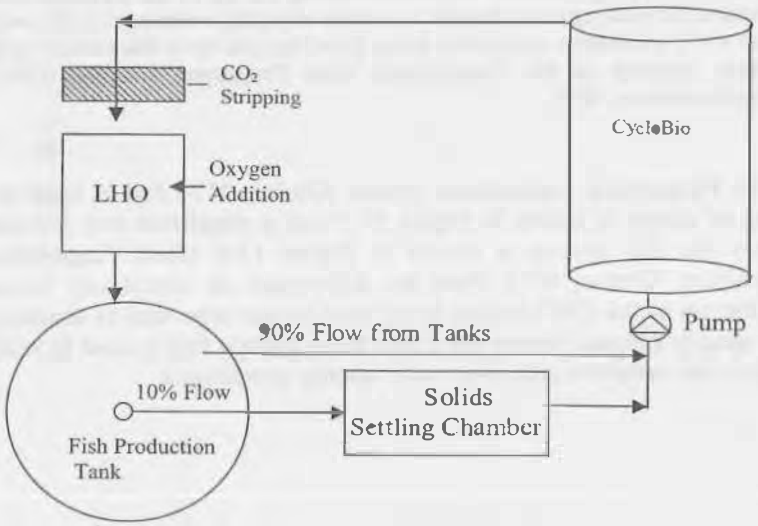

Figure 17.4 Unit process flow diagram indicating CycloBio location within a Fingerlakes Aquaculture Production 'Pod' used to rear tilapia in a RAS.

COST OF PUMPING

A primary disadvantage of RAS technology is that water must be moved from the culture tank to the different unit processes that restore used water to acceptable levels of quality for fish growth. Table 17.3 summarizes the costs associated with pumping based upon the total dynamic head required (TDH, how high the water must be elevated above the culture tank free water surface plus the energy needed to overcome any friction losses in the pipe and to provide any needed pressure at the new height) and the flow rate required to support fish growth. As a rule-of-thumb, one can use 8 Lpm of flow per kg of feed fed per day (5 gpm per pound of feed fed per day) (for supplying oxygen and required nitrification). Table 17.3 illustrates the cost of pumping using various flow rates and water lifting heights.

> **"Rule of Thumb"**
> 8 Lpm of flow per kg of feed fed per day
> (5 gpm per pound of feed fed per day)

Table 17.3 Cost of Pumping per Unit of Fish Produced (kg) Assuming Electricity, \$0.10 per kWh, Efficiency of Pump @ 70% and Feed to Gain Ratio of 1.00. Note: BHP = Q x TDH x SG / 0.012 / Pump Efficiency where BHP = kW and Q = m^3/s, and TDH = m. *Assumes that each flow rate listed is supporting 1 kg of feed fed per day

Lpm/(kg feed/day)	TDH m	BHP (kW)	kWh/kg	Cost/kg (whole fish)
5	1	0.001	0.028	0.003
10	1	0.002	0.056	0.006
20	1	0.005	0.112	0.011
30	1	0.007	0.168	0.017
40	1	0.009	0.224	0.022

For example, if the TDH needed to run your particular RAS system were 5 m, e.g., for a fluidized sand bed, and the feed to gain ratio for the system is 1.5 kg feed per kg gain, then the cost of pumping assuming a flow of 30 Lpm per kg of feed being fed per day would be:

$$\frac{Cost}{lb} = 0.168\frac{kWh}{m \cdot kg_{feed}} \cdot 5m \cdot 1.5 fg \frac{kg_{feed}}{kg_{fish}} \cdot \frac{\$0.10}{kWh} = \frac{\$0.126}{kg_{fish}}$$

Thus, it can be seen that production costs for pumping are proportional to:

- Pumping pressure (total dynamic head the pump works against, TDH)
- Feed to gain ratio (fg)
- Electricity cost

Design and planning should address lowering all three of these contributing factors to pump operating costs during the first stages of RAS farm planning. Design considerations should include how to incorporate low head pumps and water treatment methods that minimize requirements for lifting water.

BIOFILTRATION

Effective and cost efficient biofiltration is one of the keys elements to cost effective indoor aquaculture production. The choice of biofilter will impact the dynamic head that the pump system must work against. Fluidized sand beds will work against 5 to 10 meters of head, while trickle type filters can be operated at much shallower depths, e.g., 2 m. Floating bead biofilters are similar in head requirements to trickle filters and provide large surface areas for nitrification comparable to fluidized sand beds. All biofilters have advantages and disadvantages, and for small scale systems, e.g. feeding around 50 kg of feed per day, the choice of biofilter is probably irrelevant. Based upon the authors' experiences, we summarize costs for various biofilter choices (see Table 17.4) based upon their capitalization cost to support a 454 ton per year tilapia farm (1 M lb/yr).

Table 17.4 Capital Costs Estimates Associated with Biofilter Choices for a Tilapia Farm Producing 454 M ton (1 million lb) Annually. Cost is listed as $ per kg per year of production capacity. Not all the biofilter types described in Chapter 7 are included here, and inclusion here does not constitute a recommendation.

Biofilter Type	Farm Cost	Cost, $ per kg/yr
Rotating Biological Contactor	$668,000	$1.50
Trickling Biofilter	$620,000	$1.36
Bead Filter (not microbead Aquafilter type; see note below)	$296,000	$0.66
Conventional Fluidized-Sand Biofilter	$124,000	$0.26
CycloBio™ Fluidized-Sand Biofilter	$76,000	$0.18

Trout, char, and salmon require relatively clean water and low levels of un-ionized ammonia, so high percent removal efficiencies per hydraulic pass are required when designing systems to raise these species. For this reason, fluidized-sand biofilters containing fine sands are commonly used in cold-water recirculating systems because these biofilters will often achieve 70-90% TAN removal efficiencies. Fluidized-sand biofilters using fine sands are also capable of providing complete nitrification (due to their excess supply of surface area), which helps to maintain low nitrite-nitrogen concentrations within the recirculating system (generally < 0.1-0.2 mg/L as nitrogen). As can be seen from this discussion, the choice of species will affect the choice of equipment and biofiltration technology employed. The costs of production as affected by capitalization and water quality maintenance must be kept in the forefront if RAS technology is to be applied on an economically successful basis. It is a relatively easy design task to develop a system that will create high water quality conditions, but designing a system that can produce fish economically is an entirely different design task and is in fact quite challenging.

Gas Stripping and pH Control

RAS systems can require pro-active design components to remove dissolved carbon dioxide and to supply sufficient oxygen to maintain fish productivity. A typical gas transfer device that combines both functions is shown in Figure 17.5.

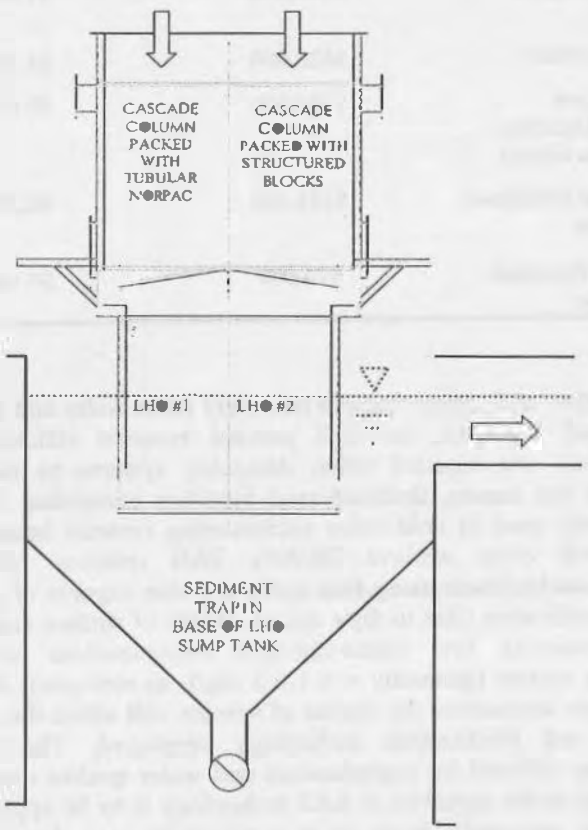

Figure 17.5 Stacked CO_2 stripper/LHO unit supported over a cone bottom sump tank to simplify sediment removal. (drawing by PRAqua Technologies Ltd., Nanaimo, British Columbia, Canada).

Controlling CO_2 stripping rates can be used to control water pH and pH control can be used to mitigate problems associated with high ammonia. These unit process devices are continually being improved for practical implementation.

Solids Removal

The effective control of solids in RAS is probably the *most critical parameter* for long term economic success. Poor solids removal destroys water quality and hence fish performance and compromises biofilter performance. Currently the most generally used method for solids removal is mechanical screens using 60 to 120 micron mesh sizes. Unfortunately, mechanical screening for solids removal is typically now the most expensive single component of an entire RAS system. Settling basins with frequent cleaning (e.g. twice per day) may be appropriate where labor costs are relatively low in less developed countries. Design details for solids removal are provided in Chapter 5 Solids Capture.

Other Costs

Feeding systems, alarm and control systems and all other capital cost components are often the same equipment being used on outdoor or flow-through non-RAS systems. Consideration should be given that a different environment can exist within RAS farms that are generally indoors, e.g. high humidity. Building construction, insulation materials, heating and ventilation systems – must all be considered to create an effective farm design, see Chapter 14 Building System Environmental Control. For example, using wood structural components in indoor RAS environments to save on initial costs will have long term effects, since these type structures will need to be replaced typically every 3 or 4 years or so. Using wood can be a good idea, though, when you are not sure of your design and you expect to be changing components over a fairly short period. Don't over invest in experimental designs, except to provide realistic functionality of a test concept.

17.7 Scale Effects and Risk

The primary consideration that should dictate a particular biomass fish load for an independent production system is associated with catastrophic failure. Systems fail as units, and typically all fish in a particular failed unit will die. Biofilters are one source of system failure. One approach is to failure mitigation is to minimize the number of

biofilters on a farm so that each can be designed with substantial redundancy and excess capacity, thus minimizing risk in this regard. The alternative approach is to build several small biomass systems, each with an individual biofilter, so that when a system fails, the severity of the economic loss is small since an individual system represents only a small fraction of the total production capacity. The FLA farm was designed with six independent production systems. Each production system was capable of producing 250,000 lb (114,000 kg) per year of fish; thus system components within a module were designed to handle this biomass load and associated feeding rates. By designing the farm as six independent units, this means that a particular unit could fail without impacting the other five units. It becomes an issue of distributing risk and risk associated with a system failure versus the economies of scale associated with designing larger and larger production modules. Design farms with at least four independent growout systems, so that in the event of a catastrophic failure to a specific system, the farm would only lose 25% of the current crop.

Biofilter characteristics are shown in Tables 17.5 and 17.6 (from Chapter 7 Biofiltration). Note the difference in space and volume requirements between the two filter types. For small farm applications, e.g., less than 50,000 lb/yr of production (23,000 kg/yr), the choice of biofilter type probably does not make much difference. It is only in larger farms where the filter selection has a noticeable affect on overall profitability. So, if you are just starting, choose the biofilter that makes the most sense and is simplest for you to operate and service. Trickle or moving bed biofilters are often a good starting choice.

Table 17.5 Ammonia Assimilation Rates for Different Classes of Biofilters

Filter Type	Rating Basis	Biofilter Ammonia Oxidation Rates 15–20°C	25–30°C
Granular	Volume of Unexpanded Media	0.6–0.7 kg TAN/m^3/day	1.0–1.5 kg TAN/m^3/day
Trickling & RBC	Surface area of media	0.2–1.0 g/m^2/day	1.0–2.0 g/m^2/day

Table 17.6 Volumetric Requirements for the Two General Types of Filters for a Unit Nitrification Loading Factor

Filter Type	Surface Area per Unit Volume m^2/m^3	TAN Oxidized per day	Assimilation Rate	Media Volume Required
Granular	1,000 to 10,000	1.0 kg TAN per day	1.0 kg TAN/m^3/day	1.0 m^3
Trickling & RBC	150–300	1.0 kg TAN per day	1.0 g TAN/m^2/day	5.0 m^3

Economically Competitive Scale

You will have to size the scale of your operation to be competitive in your chosen market. If you are going to market your own product, this will allow you to compete at the retail level, but this marketing strategy will require a significant time commitment to serve a series of small customers. For example, a restaurant customer will usually only want 10 to 20 lbs (5 to 9 kg) of fish per week, usually in fillet form. So, get ready to start cutting up fish!

The typical large tilapia farm in the US produces between 200,000 to 1,000,000 lb (91,000 to 455,000 kg) per year of whole fish. As large as this seems to be, these levels of production are not adequate to be competitive with the overseas producers of fresh tilapia fillets. They (the overseas competitors) are able to process fillets for about $0.30/lb ($0.66 kg), and their farms produce between 5 to 15 million lb/year (2,500 to 7,500 ton). You can do some quick pencil calculations on the costs to prepare fillets that will show even if you are really good at it, your hand labor costs will still approach $1.00/lb ($2.20/kg) fillet basis. Expert hand cutters and trimmers might be able to produce a fillet product for a cost as low as $0.50/lb ($1.10/kg). Obviously, this places you at a severe processing cost disadvantage to the large overseas farms, so hand labor processing is not feasible.

Fully automated processing equipment can reduce fillet processing costs to $0.15 to $0.25 per lb ($0.33 to $0.55/kg). Processing costs at this level would allow US producers to be cost competitive with the overseas suppliers. The problem is that the automatic filleting equipment requires volumes on the order of 5 to 10 million lb (2,300 to 4,500 ton) per year to justify the initial equipment expense. Additionally, other economies of scale start to emerge at this level of production as well. You will be able to do cost-benefit tradeoffs associated with in-house feed manufacturing, oxygen production, electrical generation, and fish by-product utilization. Having a production capability in the range of several hundred thousand

pounds (45 ton) per year is probably the worst position to be in. Basically, at this level of production, you would have more fish than you could market to local niche local customers, but you would need a fairly large staff and significant fixed costs to operate the farm. All of these factors contribute to high overall production costs. Either you get really big like the successful catfish farmers have, or you stay really small and basically do everything yourself.

"Rule of Thumb"

A production level of several hundred thousand lbs (45 ton) per year is probably the worst position to be in.

17.8 LABOR REQUIREMENTS

Labor costs will become a major factor in your operation. Large farms will provide human coverage 7 days a week, 24 hr per day (24–7). Smaller growout farms rarely employ this type of coverage. The question becomes what is large and what is small. The financial ramifications of catastrophic failure answer this question. If you can economically absorb the loss of all your fish, you might consider limited coverage. Certainly, during the first year of growout operations, we highly recommend as much coverage as possible. It is simply amazing the ways that failure can occur. Catastrophic losses will rarely occur while a human is on-site checking the fish and operation of the system equipment (this on-site human presence is in addition to the automatic sensing and monitoring that is in place, see Chapter 13 System Monitoring & Control). As the system matures and built in alarm systems for loss of flow, water level, low oxygen etc. are infrequently activated, then one could consider easing the coverage of the fish farm. Experience shows that about 50% of the problems that arise during farm operations are detected by the automated alarm system. Additionally, the alarm systems will occasionally detect a problem when there is none. Yes, they should be much better, but this is what happens in the real world. There is a delicate balance between monitoring too many things and not enough, and over-reliance on automated warning systems versus having none.

The authors' experience with a variety of tanks ranging in size from 2 to 11 m in diameter is that tanks of various sizes require similar man-hours to manage. In effect, it is the number of tanks and not the size of

tanks that is important in determining management hours required. Efficient growers can adequately manage a series of tanks with a time budget of 20 to 30 minutes per day per tank system (11,000 L). Daily activities include daily water chemistry measurements, fish feeding, filter maintenance, and tank cleaning. Weekly maintenance of two to three additional man-hours per tank system for major cleaning activities and preventative maintenance is also necessary. Assuming a 40-hour work week, this indicates that one person could manage seven to nine tank systems (average of 4.3 to 5.3 man-hours per tank system per week). This is also consistent with the Fingerlakes Aquaculture experience, e.g., five line staff to manage a 48 tank facility 24–7.

Since many operations on a farm require two people, a facility could be designed assuming two full-time employees/owners to maximize labor efficiency. What is generally overlooked in estimating labor requirements is the additional short-term labor associated with intensive tasks such as harvesting and repair work. It is probably reasonable to estimate that there is about twice as much labor required as you can calculate as being needed. When everything is going well, fish systems require minimal labor, but then there is the rest of the time when things don't go so well! A farm owner might consider using hourly or contract labor for special tasks, e.g., harvesting, hauling, processing, etc. Losordo and Westerman (1994) used 8 hours per day to manage an eight tank facility with a 3 tank nursery (approximately 50% of the labor per tank used in our cost analysis).

17.9 PREDICTED COSTS OF PRODUCTION

The technology being used must support an overall economically competitive operation. The initial cost of your start up equipment will define in part the company's anticipated internal rate of return on the investment. You should assume a five year depreciation schedule for this equipment, but this factor is less important than initial costs in determining your expected economic returns. The depreciation schedule allowed by the IRS for a single purpose agriculture building is 10 years (MACR or 15 years in ADS, straight line), which is good for minimizing taxable cash flow, but terrible when you are showing rates of return on your investment. For larger scale farms (200,000 lb or 91 ton per year and larger), a good ratio of capitalization cost to production is that the equipment cost should be $0.50 to $1.00 per lb ($1.10–$2.20 /kg) per year of production and the cost of buildings, land, utilities, etc. should not exceed an additional $0.50 to $0.75 per lb ($1.10-$1.65/kg) per year.

The range is because these costs are very species and site specific. For small farming units, these costs will be at least twice these rates and could conceivably be even fourfold higher. If the up-front investment costs are too high, your farms internal rate of return on investment will be too low to attract outside investors.

Calculating costs of production, profits, and losses require a basic understanding of common economic terms. These are given for ease of reference below. The reader should consult a more complete business text for further help; e.g., Bangs, D.H. 2002: The Business Planning Guide, Publisher: Kaplan Business; 9th edition (April 15, 2002, ISBN: 079315409X. The Bangs text is a succinct description of business economics and also covers the basics of writing an effective business plan.

TERMINOLOGY FOR ECONOMIC ANALYSIS

PROFIT & LOSS STATEMENT

This is called a Pro-forma when it projects profits and losses (P&L's) into the future.

REVENUE

Self explanatory; usually set up in spreadsheet format where you can easily change price received for products sold. Create as many categories as necessary for products being sold.

Total Revenue = Sum of all revenues

COST OF GOODS SOLD (CGS)

Sometimes called direct costs, these are proportional to the amount of goods actually sold, e.g., feed, fingerlings, oxygen, processing fees.

FIXED COSTS

These costs are incurred whether you sell any product or not (often called your overhead; goal is to make Fixed Costs as small a percentage of total costs as possible). Fixed Costs can be described as:

Fixed Costs = Operating Expenses + Selling, General & Administrative (SG&A)

Operating Expenses = Electricity (pumping) + Heating (air and water) + Hourly labor (front line labor) + Supplies + Insurance (building, key man, liability) + Taxes (property, payroll, sales, NOT Income taxes) + Royalties

SG&A = Management payroll, legal fees, professional services

TOTAL EXPENSES AND NET INCOME

Total Expenses = Variable Costs + Fixed Costs

Net Income (EBITDA) = Revenue - Total Expenses

Note: at this point Net Income does NOT reflect interest, taxes on profits, depreciation or amortization; hence the acronym EBITDA (Earnings Before Interest, Taxes and Amortization). EBITDA is very useful since the ITDA of the EBITDA can be very dependent upon local banks, equity invested, etc. Financial analysis usually addresses this by calculating Internal Rate of Returns or Net Present Values.

DEPRECIATION

Depreciation is the loss in value over time of a physical asset. From a tax perspective, the IRS regulates depreciation by creating classes of items, e.g., a truck, computer, pump, building. Each item has a maximum allowable depreciation period, e.g., light equipment might be 7 years; single purpose agriculture building = 10 years; a commercial building = 39 years, a residential building = 28.5 years. Having selected the appropriate depreciation period for the class of item, then there are again IRS required methods to calculate depreciation for each year an asset is in service, e.g., straight line, double depreciation, MARCS (Modified Accelerated Cost Recovery System).

The Straight Line is the easiest and least confusing method, but accounting firms seem to like MARCS, which requires a look up table approach and accelerates depreciation to the front of the period.

Using straight line, you have to assign a salvage value to the asset or the items value when it would be finally taken out of use at the end of its life cycle. Again, the simple approach is to assign zero, which maximizes allowance depreciation each year, and then you calculate depreciation as:

$$Depreciation\ Cost(\$ / yr) = \frac{Total\ Cost - salvage\ value}{Years\ of\ life}$$

Units become \$/yr. So, if you purchased a pump for \$1,000, assigned zero salvage value, and assigned a useful life of 5 years, your cost per year for depreciation on this item is \$200/yr. In terms of government taxing, the depreciation cost is subtracted from income making taxable income less by the depreciation amount. Now, the tricky part is that if you sell the item being depreciated, this becomes income for your company if sale price is greater than book value. Net income for this sale will be selling price less your current book value of the item, which is initial value less the claimed depreciation to date. So, if you sell the item for its current book value (the value of the asset on the company's tax records), no taxable income is generated.

ECONOMICS OF RAS

Based upon our experience using RAS, the following table summarizes costs and required inputs for a commercial scale tilapia farm in a northern US climate location.

As mentioned earlier, smaller farm units will incur much higher production costs per unit, and unit input parameters will also be significantly higher. For example, purchasing oxygen in typical welding size cylinders (282 cubic feet, 8 m^3, or 23.5 lbs, 10.7 kg, of oxygen), will cost \$3 or \$4 per 100 cubic feet (2.82 m^3) of product (or 10 fold the costs listed in Table 17.6). Labor is another area where the larger farms achieve great economies of scale, since it takes almost the same labor to service small tanks as it does large tanks and it takes about the same amount of time to for security checking whether there are two tanks or 20.

OXYGEN USE & CARBON DIOXIDE PRODUCTION

In the above table, one of the more confusing things is probably how we've assigned such different rates of oxygen use for small versus large farms (1.9 for small farms and 1.0 for large farms). In fact, the stoichiometric ratio could be as low as 0.37 kg of oxygen per kg of feed fed (0.25 for fish metabolism and 0.12 for nitrification). For design purposes, using a ratio of 1.0 kg oxygen per kg of feed is a good starting point. It must be recognized that in small farms, the efficiency of use will just be poor for a wide variety of reasons, e.g., lack of qualified personnel, lack of attention to maintenance, leakage. Of course, these things could happen in a large farm, too. It is also essentially impossible to go to the theoretical lower limit as systems will always have varying degrees of suspended solids and heterotrophic activity and thus additional oxygen usage. While these solids are coming from the feed and therefore the bacteria resulting from them and the associated oxygen demand resulting from the bacteria's metabolism should already be accounted for, it just doesn't seem to work out that way. The most efficient oxygen usage in a farm of any scale that we've seen is around 0.5 kg of oxygen use per kg of feed fed.

Regardless of how much oxygen your system ends up using, the carbon dioxide production will be 1.375 times as much, since this is a stoichiometric relationship. A flow-through system would therefore have an advantage in this regard, since there would be less oxygen consumption because no significant nitrification is occurring and hence less oxygen consumption per unit of feed being fed.

FEED AND OTHER COMMENTS

In the table below, you also see how we have discounted the feed price for the large farm. It is the same feed, why the difference? Once you have a large farm, the feed will be delivered in truck load quantities and augured or blown directly from the feed truck into your feed bins at your farm. This efficiency plus your ability to command a lower price by the bidding process to various feed suppliers should result in the price reductions shown above. And back to the oxygen discussion, you will also receive discounted oxygen rates once you reach a truly competitive size at around the 2,500 ton/yr farm size, and at this large scale, you will probably ***go into self generation of*** oxygen using similar equipment as what the oxygen supply companies employ. This will result in roughly a 25% reduction in your cost or more depending upon your electrical cost rates (the gas purification process uses electricity to run the separation process).

Table 17.6 Unit Costs and Required Inputs for Large Scale Tilapia Production

Item	500 ton/yr farm (1 million lb/yr)	2,500 ton/yr farm (5 million lb/yr farm)
Size of Building, ft^2 (m^2)	40,000 (3,720)	200,000 (18,600) Proportional 5x
Total Cost of Facility & Equip	$1,500,000	$7,500,000 Proportional 5x
Number of Pods (replicated production units, see Chpt 5)	6	30 Proportional 5x
Volume per Pod unit, gallons (m^3)	76,000 (288)	same
Production per Pod, lb/yr (kg/yr)	250,000 (114,000)	same
Direct Labor Per Building	5 FTE	20 FTE
Growth rate: egg to 700 gram	28 weeks	same
Feed Conversion Ratio (high quality high protein feed)	1.0 to 1.2	same
Electric Use, kWh per Lb (per kg)	1.3 (2.9)	same
Heat Use, Therm[b] per lb (kJ/kg)	0.17 (48,000)	same
Oxygen Use lb/lb (kg/kg) of Feed Fed	1.9 (low efficiency)	1.0 (high efficiency)
Feed Purchase Cost (41% Protein) $/lb ($/kg)	$0.28 ($0.62)	$0.18 ($0.40)
Oxygen Supplied cost[a], $/100 ft^3 ($/m^3)	$0.34 (0.12) + facility fee[a]	same
Electrical Power, $/kWh	$0.04	same
Gas Cost, $/Therm ($/100,000 kJ)	$0.85 ($0.80)	$0.10 ($0.095) (energy plant)[c]
Unit Cost: $/lb whole fish (fillet basis)	$0.78 ($2.52)	$0.55 ($1.77)
Process, Pack, Ice $/lb (kg) fillet	$1.50 (3.30)	$0.55 (1.21)
Cost of Fillet FOB, $/lb (kg)	$4.02 (8.84)	$2.32 (5.10)

[a]Facility fee will usually add around 20% to the cost of the delivered oxygen
[b]Therm = 100,000 BTU (105,500 kJ)
[c]Large scale farms would most likely be placed at a site with an available, underutilized heat source

17.10 PREDICTED COSTS OF RAS PRODUCED TILAPIA

A cost analysis for a generic tilapia RAS based farm is shown in Tables 17.7-9 (software available at www.bee.cornell.edu/aqua). Data in Table 17.7 and 17.8 is considered representative for a current state-of-the-art tilapia farm producing in excess of 1,000 ton/year (2 million lb per year). The predicted costs (Table 17.9) of production for such a farm indicates that a large commercial tilapia farm could compete quite effectively with offshore production in the fillet fresh market *if processing costs* can be achieved competitively (a major challenge still). A series of 1,000 ton (multi-million lb) production farms would be required to support an automated processing facility.

Table 17.7. Inputs for Large Farm Model in English units (2 million lb/yr)

Building Costs, Maintenance and Labor					
Days of Operation Per Year		Total Bldg floor area square feet	Number of Pods in Building	Gallons in Tanks, Sump, Biofilter per Pod	Complete Pod system w/access, $/pod
365		60,000	8	100,000	$125,000
Equip. depr. period, yrs	Building depr. period, yrs.	Total Labor Cost $/yr-building	Initial Cost of Building	Building Backup and Monitoring Costs	
7	12	$200,000	$600,000	$50,000	

Fish needs (oxygen, feed and fingerlings)					
O2 Cost $/lb	lb O2 supplied per lb of feed fed	Feed Cost $/lb	Feed to Gain Ratio	Min Feed lb/day-pod	Max Feed lb/day-pod
$0.04	0.75	$0.14	1.20	400	822
Mortality over growth period	Cost $/fingerling	Selling Size lb			
5.0%	$0.05	2.00			

Heating and Electricity					
Water temp deg F	Temp makeup water, deg F	Daily Water Exchange Rate	Heat Exchanger Efficiency	Overall R value for Building, hr*ft^2*F/Btu	Air Volume Changes, ft^3/hr
82.0	50.0	10.0%	0.0%	20.0	2.0
Cost of Energy $ per 100,000 BTU	kW per pod system	Electrical Cost $ per kWh			
$0.75	12.0	$0.04			

Note: Oxygen efficiency of use is reflected in the supplied oxygen level per unit of feed fed
Note: Assumes heating of air and water is based upon cost of energy shown

Farm Output, lb/year 2,000,000

Table 17.8 (below) gives design parameters that can be used when trying to predict RAS performance. The actual values used will be very design dependent and highly impacted by the management skills of the RAS team.

Table 17.8 Production System Characteristics Associated with Tilapia Indoor System

Production Characteristic	Value
Size of building	1,000 to 4,000 m^2
Individual growout tank (a set will make up a production pod)	16 tank facility (7.6 m diameter x 1.4 m deep) 60,000 L
Yearly harvest	500 ton and larger
Design Parameters	
Harvest Density	100–120 kg/cubic meter
Feeding rate (depends on fish size)	2% to 3% body mass per day
Feed conversion rate (feed/gain)	1.00–1.40 kg/kg
Supplemental oxygen	0.4–1.0 kg oxygen/kg of feed fed
Oxygen absorption efficiency	75%
Power per tank system	Extremely design dependent
Fish target harvest size	680g– 1.0 kg
Daily water discharge, % of system volume	5–20%
Temperature difference for water exchange	Site dependent
Building infiltration, minimum air volumes/hr	2–3

Table 17.9. Predicted Production Costs for a 1,000 ton (2-Million lb) per Year Tilapia Farm (costs in \$/kg) for Inputs from Table 17.7-8

Feed/Pod (kg/day) & 8 pods	374	
Production (kg/week) (whole fish)	17,045	% Total
Electric Cost	\$0.04	3%
Feed Cost	\$0.37	30%
Water Heating	\$0.07	6%
Air Heating	\$0.07	6%
Oxygen	\$0.09	7%
Labor	\$0.22	18%
Fingerling	\$0.06	5%
Depreciation & Repairs	\$0.31	25%
Total Cost (\$/kg)	\$1.23	100%

The capitalization cost for the above example is \$1.83 per kg per year of production capacity. Labor costs are relatively high compared to salmon farming. In comparison, Atlantic salmon capitalization costs for 10 to 20 net pens per site are \$50,000 per pen (good for stocking 50,000 smolts), the capitalization cost expressed based upon system production capacity per year is:

\$/ (kg per year capacity) = \$500,000/ (1,300,000 kg/year) = \$0.40/kg/yr

Note that the tilapia capitalization (**\$1.83/kg/yr**) is over four times larger than the salmon capitalization. Table 17.10 summarizes the impact of capitalization costs (investment) upon costs of production. The salmon costs are by far the lowest in the aquaculture industry and this can be attributed to its large scale farming approach and a concerted application of research to provide the needed equipment and management techniques for this industry.

Table 17.10 Depreciation Cost (\$/kg) Fish Produced as Affected by Depreciation Period (straight line)

	Depreciation Period, Years		
Capital Cost \$ per kg/yr	4	7	10
\$0.40	\$0.10	\$0.06	\$0.04
\$0.50	\$0.13	\$0.07	\$0.05
\$1.00	\$0.25	\$0.14	\$0.10
\$2.00	\$0.50	\$0.29	\$0.20
\$3.00	\$0.75	\$0.43	\$0.30

Capitalization Components

Table 17.11 below illustrates how the total capitalization costs for a large scale tilapia farm are distributed over a wide range of required components. Percentages could be very different for a small-scale farm, e.g., < 25 to 50 ton/year. Note that the growout systems themselves (tanks, water quality control, feeding systems, and hatchery area) only account for 32% of the total cost of the project (separate component costs are not broken down here, since typically a "system" is purchased that addresses the different unit process needs). Many new people looking into the business think that if they have free tanks, then their capital costs to start a fish farm will be modest. Note that the fish tanks are only 3–5% of the 32% (or 1/10 to 1/5 of the 32%) estimated for all the equipment. Many people will fail to include many of the support components needed

in a fully equipped farm and as a result, grossly underestimate the amount of capital needed to start a fish farm.

Table 17.11 Capital Cost Characteristics Associated with a Large-scale Tilapia Water Reuse System, e.g., 600 ton/yr.

Component of Initial Capital Cost	%
Production Tank Systems (components not broken down, only identified)	
Growout tanks, pumps	
Oxygen and CO_2 control units	
Electronic controller	
Feeders	
Large Scale Biofilter (treat multiple tanks)	
Quarantine hatchery /fingerling area (series of small tanks)	
Subtotal tank costs (approximate)	32%
Other Equipment (approximation)	
Backup generator (400 kW system)	5%
Monitoring system	2%
Ice machine (2 ton unit)	1%
Feed bin and auger system	2%
Harvesting system	2%
Water heating system	2%
Waste catchment unit	1%
Ventilation system	1%
Water wells (2)	1%
Fish handling equipment	1%
Subtotal Other equipment	18%
Total Equipment Costs (7 year depreciation period)	**49%**
Building Costs	
Quarantine area	1%
Laboratory and office space	1%
Building space	46%
Septic/restroom	0%
Subtotal building Costs (10 year depreciation period)	**49%**
Land costs (non depreciable)	2%
Total funds required (equipment, building, land and contingency)	100%

Other Start Up Costs

Before we depart this subject, we should also point out that in properly capitalizing your project, you must include adequate working capital in your fundraising efforts. You will need to build inventory before you can sell your first fish. Thus, you need enough funds to cover all the expenses incurred before selling the first crop. You should also anticipate some delay in bringing your first crop to market (you should probably be targeting a weekly harvest for sales throughout the year to maximize the benefits that can be obtain using RAS technology) for a wide range of reasons. Remember that you will have fixed costs that just keep on draining your cash reserves even if you are not feeding the fish, e.g., labor, rentals, management overhead—or collectively what we refer to as SG&A: Sales, General, & Administrative expenses. Finally, once you sell your product, it may be as much as two months before you receive the cash for the goods sold. **You cannot run out of cash**, so be sure to keep these additional cash needs in mind from the beginning of your planning process.

17.11 Economic Comparison to Broilers and Catfish

The authors admit that we are enthusiastic about the economic prospects of RAS technology. An often encountered argument against RAS is that they can not compete against large commercial fish ponds such as those used by the US commercial catfish industry. The short answer to argument is that YES, fish produced in using RAS can compete against fish produced from traditional rearing systems, e.g., ponds, net pens, raceways. In the 1st and 2nd editions of this text, we provided a case analysis comparing tilapia produced using RAS to catfish from southern USA pond systems (or see Timmons and Aho, 1998 for the same set of tables). The data for the catfish farm was from a 1988 study (Keenum and Waldrop, 1988) so we felt this was a bit dated for the current text. However, our analysis showed that the key to the RAS system being competitive was that the tilapia RAS farm had to be on the same scale as the catfish farm. Pond systems have serious limitations that compromise their economic productivity, such as:

- Must be located in a warm climate to allow high productivity, e.g., 5 to 10 tons per hectare
- Require significant investment in land cost due to their extensive nature (compared to RAS)

- Labor requirements are higher than initially one might think due to the maintenance of a large land area/pond bank
- Feed conversion efficiency is poor compared to RAS where ideal water environments can be maintained continuously throughout the year
- Vulnerable to disease issues/catastrophic weather events

We also compared both the RAS tilapia and catfish pond systems to USA broiler production costs (again see Timmons and Aho, 1998). What this showed was that broiler production is still immensely cost superior to both catfish or RAS produced tilapia (total absorbed costs for broiler production are ~ $0.65/kg for a pre-processed whole bird).

Ultimately, fish production from aquaculture will have to compete with other commodity meats such as poultry. As just pointed out, costs of broiler production in the USA are extremely efficient (~ $0.65/kg whole bird, preprocessed). The broiler industry should serve as an inspiration to all of us of what can be accomplished when farmers and agribusiness work together towards a common goal. USA broiler production is a vertically integrated industry, with the broiler grower being the contract farmer and the foundation of the system. The farmer owns the building, provides husbandry, and pays most of the cost of utilities. For these services, the farmer is paid approximately $0.09 to $0.11 per kg of broiler produced. Thus, all costs associated with building ownership, depreciation of capital equipment, labor, and utilities (electric and water and generally about 50% of the fuel heating costs) are borne by the farmer. The productivity per worker has increased from 95,000 kg of broilers per year in 1951 (Watt Publishing, 1951) to 950,000 kg per year in 1991 (Perry, 1991). According to Dr. Paul Aho (Poultry Perspective, Storrs, CT), current productivity (2006) has increased even further to ~1,300,000 kg per year per full-time worker. Wow! Aquaculture production does not even approach these productivities....yet. (See Chapter 1 for more discussion)

The broiler industry has seen a continuous increase in improved efficiency and bird performance for equipment, housing, and nutrition and genetics. North (1984) provides an extensive description of all facets of commercial poultry production. It is interesting to note that broiler production in the 1950's was around 5 million kg per year. The productivity per unit of worker and total broiler consumption of the 1950's is very similar to the current productivity standards of the USA tilapia industry (8 million kg per year whole fish basis) and the productivity per person in the fish farming business is approximately 25,000 to 110,000 kg per year.

The primary factor that led the poultry industry to convert from outdoor production to indoor farming was the tremendous savings in labor costs. As mentioned earlier, the labor productivity for indoor broiler production is currently about eight times greater than the labor productivity currently being obtained for indoor tilapia fish farming. Ultimately, indoor fish production has two distinct advantages over poultry production: higher feed conversion efficiency and greater productivity per unit area of building. Broiler production has feed conversion efficiencies of 1.95 (2.09 kg bird weight, feed to gain ratio on feed energy levels of 3,170 kcal/kg and protein levels of 19.5%), while tilapia conversions are currently in the 1.2 to 1.5 range for feed energy levels of approximately 2,500 to 3,200 kcal/kg and protein levels of 32 to 40%. The yearly meat output per unit floor area from the tilapia system is 255 kg/m^2 (and it can be doubled with good engineering and management) compared to 122 kg/m^2 from a broiler house. Thus, net economic productivity per year from a fixed tilapia production facility could be much higher than that obtained from a broiler facility of similar size, even though the unit costs of tilapia production per weight are slightly higher in comparison to broilers. The advantage for fish production systems is their higher potential rate of return per year from a fixed facility. (More discussion on this topic is provided in Chapter 1)

FUTURE EXPECTED IMPROVEMENTS IN COST EFFICIENCY

At this point, there is considerable information available regarding the expected cost of tilapia production for large scale operations and what kind of labor savings could be anticipated to develop over the next ten years. For example, many public electric utilities will reduce electrical rate charges by 25% once a load of 500 kW is reached. Large facilities can self-generate electrical power using large electrical generators. This is particularly attractive, since a generator system is required whether the generator is used as a backup system to power outage or as a primary generator. In this case, the cost of electricity to the fish farm is only the operating cost of the unit. Maintenance and depreciation will be slightly more when the generator is used as primary as opposed to backup, but there will not be a lot of difference. Labor requirements will continue to reduce due to automation and tank scaling and we use 50% of the current example in our projected cost scenario. Catastrophic fish insurance would no longer be deemed necessary, since the farmer would become self-insured. System costs are expected to decrease by 25% in comparison to current costs due to improvements and refinements in system designs.

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Chapter 18

Fish Nutrition And Feeds[1]

18.0 Introduction

This chapter is an overview of fish nutritional requirements and the general physiological considerations that influence the nutrition, feeding, and welfare of fish. It is not meant to be a definitive nutritional text; the intent is to provide basic information to non-nutritionists so they might be able to better understand the inter-relationships between the Recirculating Aquaculture System (RAS) and the cultured animals.

18.1 Feed Management

Feed management involves selecting the proper feed, ordering the proper amounts, scheduling the delivery in a timely manner, storing the food properly, using older feed first, and implementing efficient feeding methods. All of these factors can have an effect on the bottom line. Poor feed management can result in any number of problems. For example, if the proper feed size or formulation is not selected, the fish may not grow efficiently. If a low-phosphorus feed is not selected, effluent problems can result. On the other hand, sound feed management practices can assure optimum growth and lower total costs. The purpose of this section is to discuss the steps needed to properly acquire, store, and distribute feed. Also, for those with limited background in fish nutrition, we review some basic principles of feed nutrition.

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FEEDING TECHNIQUES

The goal of efficient feed management is to calculate the proper ration and present it to the fish. Underfed fish do not reach maximal growth, and may exhibit aggressive behavior during feeding due to limited feed availability, thereby potentially harming themselves or other fish. Underfeeding may also increase the variability of fish sizes within a tank. Overfeeding results in uneaten feed that results in poorer water quality, poorer economic performance, and additional environmental pollution.

METHODS

As a percentage of body weight, the quantity of feed required to sustain optimum growth will decrease as the fish grow from fry to fingerlings (see Table 18.1). The fish need to be fed frequently when they are small. By using belt feeders, you can feed continuously or at selected periodic intervals. Generally, the ration for the fish should be the amount they can eat in the 15 to 20-minute period after feed distribution. Crampton et al. (1990) presented an equation to suggest interval times between feedings for salmonids based upon the rate of digestion. Equation 18.1 can be used to develop a scheduled feeding program. As can be seen, the frequency of feeding can be decreased as the fish gain body size. Equation 18.1 should not be applied below 6°C and some maximum interval should be applied, e.g., 24 hours.

$$INTERVAL = 4.0\frac{\sqrt{W}}{T^{1.1}} \tag{18.1}$$

where

INTERVAL = time between feedings, hr
W = body mass, g
T = temperature, C

Often some feed may exit the tank before the fish are able to consume it. If this occurs, feed more slowly over a longer period of time, or deposit feed further from the drain so that the feed takes longer to exit the tank. A rule of thumb for the quantity of ration is to feed a weight of 1% or less of the biomass of the tank at each feeding interval. Feeding multiple times per day increases feed acquisition and improves feed conversion efficiency. A general guide to feeding rainbow trout is given in Table 18.1. This guide relates the amount fed to the size of the fish and water temperature.

When presenting feed, scattering food is generally better than fixed point feeding, as more of the population has simultaneous access to the feed. This helps reduce wastage, and may also increase uniformity of fish growth. For this reason, when using demand feeders, supplement with hand feeding. Demand feeders are useful because they are inexpensive devices, and make food available to the fish during times when the operator is not at the facility, such as at night. Always do some hand feeding each day for each tank, even when fully automated feeding systems are in place. The visual observation of feeding behavior is extremely important in maintaining a clear understanding of current fish health and whether or not a tank of fish is being over or underfed.

WHEN NOT TO FEED

Although the fry and fingerlings are normally fed frequently to maximize growth, there are certain conditions that require withholding feed. Take fish off feed completely if any of the following occurs:

- High temperatures
- Fish are sick or stressed
- 24–48 hours before transport
- 24 hours before sampling
- 3–4 days before processing
- Low oxygen levels
- Poor water quality

18.2 SELECTION

Simply stated, optimizing your feed costs would involve minimizing your feed costs per pound of fish growth. A variety of factors will influence this ratio, and the feed with the lowest purchase price may not necessarily result in the best fish growth rates or feed conversion. Some of the factors that will influence how feed should be selected are:

- Growth performance
- Feed quality
- Delivery costs
- Physical characteristics
- Formulation
- Impact on water quality

Table 18.1 Recommended Amounts of Dry Feed for Rainbow Trout per Day, Given as Percent Body Weight, for Different Size Groups Held in Water of Different Temperatures (or Pounds Feed per 100 Pounds of Fish), in Relation to Fish Size and Water Temperature, (Speece et al. 1982).

Water Temperature (°F)	Number of Fish per Pound: 2,542+	2,542-304	304-88	88-38	38-20	20-12	12-7	7-5	5-3.5	3.5-2.5	under 2.5
	Approximate Size in Inches: Under 1	1–2	2–3	3–4	4–5	5–6	6–7	7–8	8–9	9–10	10+
44	3.8	3.1	2.5	2.0	1.5	1.3	1.0	0.9	0.8	0.8	0.6
45	4.0	3.3	2.7	2.1	1.6	1.3	1.1	1.0	0.9	0.8	0.7
46	4.1	3.4	2.8	2.2	1.7	1.4	1.2	1.0	0.9	0.8	0.7
47	4.3	3.6	3.0	2.3	1.7	1.4	1.2	1.0	0.9	0.8	0.7
48	4.5	3.8	3.0	2.4	1.8	1.5	1.3	1.1	1.0	0.9	0.8
49	4.7	3.9	3.2	2.5	1.9	1.5	1.3	1.1	1.0	0.9	0.8
50	5.2	4.3	3.4	2.7	2.0	1.7	1.4	1.2	1.1	1.0	0.9
51	5.4	4.5	3.5	2.8	2.1	1.7	1.5	1.3	1.1	1.0	0.9
52	5.4	4.5	3.6	2.8	2.1	1.7	1.5	1.3	1.1	1.0	0.9
53	5.6	4.7	3.8	2.9	2.2	1.8	1.5	1.3	1.1	1.1	1.0
54	5.8	4.9	3.9	3.0	2.3	1.9	1.6	1.4	1.3	1.1	1.0
55	6.1	5.1	4.2	3.2	2.4	2.0	1.6	1.4	1.3	1.1	1.0
56	6.3	5.3	4.3	3.3	2.5	2.0	1.7	1.5	1.3	1.2	1.0
57	6.7	5.5	4.5	3.5	2.6	2.1	1.8	1.5	1.4	1.2	1.1
58	7.0	5.8	4.8	3.6	2.7	2.2	1.9	1.6	1.4	1.3	1.2
59	7.3	6.0	5.0	3.7	2.8	2.3	1.9	1.7	1.5	1.3	1.2
60	7.5	6.3	5.1	3.9	3.0	2.4	2.0	1.7	1.5	1.4	1.3
61	7.8	6.5	5.3	4.1	3.1	2.5	2.0	1.8	1.6	1.4	1.3
62	8.1	6.7	5.5	4.3	3.2	2.6	2.1	1.8	1.6	1.5	1.4
63	8.4	7.0	5.7	4.5	3.4	2.7	2.1	1.9	1.7	1.5	1.4
64	8.7	7.2	5.9	4.7	3.5	2.8	2.2	1.9	1.7	1.6	1.5
65	9.0	7.5	6.1	4.9	3.6	2.9	2.2	2.0	1.8	1.6	1.5
66	9.3	7.8	6.3	5.1	3.8	3.0	2.3	2.0	1.8	1.6	1.6
67	9.6	9.1	6.6	5.3	3.9	3.1	2.4	2.1	1.9	1.7	1.6
68	9.9	9.4	6.9	5.5	4.0	3.2	2.5	2.1	2.0	1.8	1.7

18.3 Growth Performance

Being able to specifically evaluate growth is the key to evaluating different feeds. Feed to gain ratio (FG) or feed conversion ratio (FCR) and growth rate are two of the best tools for evaluating the growth of your fish. FG is normally expressed as kilogram of feed per kilogram of gain and measures how efficiently the fish are converting the feed into body mass. The goal is to get this number as low as possible. In aquaculture, FG's of less than one can be obtained, especially when using high energy feeds and concurrently providing good management and water quality. Some are confused when this number drops below one, but this is possible because a large portion of fish weight is water weight (approximately 75–80%). This less than one FG ratio never occurs in warm-blooded animals; the best feed converters are chickens and their FG is around 2.0. One weakness of measuring FG is that the number for kilogram of feed fed cannot be accurately adjusted to account for the amount of feed presented to the fish but not eaten by the fish.

The rate of growth, expressed in weight or length gained per day, or Standard Growth Rate (SGR), is a unitless measurement of growth for measuring how fast fish grow. Using a more expensive feed that gives faster growth rates and/or better conversion may reduce overall costs by decreasing the total cost of feed, energy and labor required for operating the facility because the production cycle is shortened. Calculating the total costs to raise a pound of fish will show how feed selection can affect other costs. Calculating calorie and protein intake and monitoring fish mortality and disease symptoms are other aspects of benchmarking growth.

To optimize growth, consult the manufacturer's feeding tables, sample data, and use your personal observations. It may be difficult to find feeding tables that give practical instruction on quantities and schedules for feeding. If you are unable to find such a table, you may use the feeding tables provided as examples (see Table 18.1) and the growth equations presented in Chapter 3 to construct an initial feeding regimen, and then collect accurate feed and growth data to be used in refining your feeding tables for reference during future production cycles. By developing customized feeding tables, the feeding regimen will better fit the conditions at your facility. Once you have established rates of growth at your own facility (preferably as cm per month at specific temperatures), you can adjust your feeding charts to show feeding requirements as a function of temperature.

18.4 Feed Quality

Feed quality directly influences growth and health of the fish as well as the characteristics of the discharge stream. Although feed quality will vary between manufacturers, it can also vary from lot to lot from the same manufacturer. The exact formulation may change due to price fluctuations or availability of fish ingredients. Save (by photocopy) feed labels so that if you have a problem, you can revert to them as an aid to problem investigation. Although many of the factors regarding feed quality will not be known until after the feed has been tested, there are several quality related factors that can be evaluated before feeding, including date of manufacturer, phosphorus levels, and amount of fines. Compare the date of manufacture to the date of delivery. An example of a lot number stamp is given below. Ideally, the two dates should not differ by more than about a month, and all feed of a specific type should have a similar date of manufacture. Monitor delivery times, as feed sitting in a non-air conditioned warehouse or hot delivery truck can degrade feed quality.

Example: Deciphering Lot Numbers on a Bag of Feed

(from Zeigler Brothers, Gardners, PA)

LOT # 051306 066 1546

- The first set of six digits consists of three 2-digit numbers corresponding to the date of manufacture as month-day-year. In the example, the date is May 13, 2006.
- The second set of digits (max of 4 digits) corresponds to the bag identification number for that day. This is bag #66 produced on that day.
- The third set of digits indicates time of production in military time. The bag of feed was produced at 3:46 p.m.

"Fines" are extremely small particles of feed, and may be observed when feeding the fish. These small particles are not utilized efficiently and not only are they wasted food; they contribute to water quality problems. With current feed milling and manufacturing technology, there should be little or no fines in the bag, e.g., less than 1% fines by weight in a bag of feed. Fines are produced by poor manufacturing processes or excessive handling of the bagged feed. Feed of a certain pellet size may

exhibit fines, while feed from the same manufacturer of the same formula but having a different pellet size may be free of fines. Fines contribute to a poor quality effluent by increasing the level of suspended solids in the water column. Consult the manufacturer if there is a problem.

18.5 Physical Characteristics

Generally, feed manufacturers have three categories of feeds: starter, fry, and growout. The feed formulations optimize the nutritional characteristics and feed sizes for specific stages of fish development. The general rule for choosing a feed size is to deliver the largest size that the smallest fish will eat. The proper feed size can be selected by comparison of feed size to mouth gape size, gill-raker spacing, and esophagus width. Many manufacturers use fish length as an indicator of proper feed size for specified species. These correlations can be used to determine when to change feed sizes, but generally, observation is also an effective method of assessing when to introduce larger sized feed. In the early stages of development, the fish will be progressing to larger feed sizes rapidly, so purchase only a few bags of the smaller sized feed, and order a variety of sizes ahead of time. Use past experience in making decisions on how much feed of a particular size to order. Consider feeding behavior if selecting between sinking, floating, or slow-sinking pellets. Table 18.2 shows some recommended feed sizes as related to trout size and would be a good starting point for other species of fish.

Table 18.2 Recommended Feed Sizes as Related to Fish Size

Feed Form	US Screen Size (Mesh)	Feed Size mm	Feed Size inch	Fish Size grams	Number per pound
Starter	30–40	0.25		<0.23	2,000*
No. 1 Granule	20–30			0.23–0.6	2,000–800
No. 2 Granule	16–20			0.6–1.8	800–250
No. 3 Granule	10–16			1.8–4.5	250–100
No. 4 Granule	6–10			4.5–15.0	100–30
Pellet		3.2	1/8	15–45	30–10
Pellet		4.7	3/16	45–454	10–1
Pellet		6.3	1/4	>454	<1

Finished feeds are periodically tested to assure that tag specifications are in accordance with national regulations. In the United States, quality control procedures are in place to verify that national standards established for both feed ingredients and finished feeds are met. In this way, feed tags provide some assurance regarding the freshness and quality of feeds. Additionally, the feed manufacturer should periodically monitor feed composition materials and finished products for non-nutrient components such as Mycotoxin, thiobarbaturic acid, histamine. Vitamin E and vitamin C are highly susceptible to oxidation and should be periodically assessed. The fish farmer upon receiving each shipment of feed should (must) take and keep multiple samples (with the feedbag tag included) from every lot of feed. Feed samples must always be kept in a freezer until the cohort of fish that has consumed this food has been sold. If a problem occurs during the growout period, the feed may need to be analyzed by a reputable laboratory to determine if there may be nutritional deficiencies.

The wholesomeness of a finished feed is dictated by how well it is stabilized to retard spoilage using antioxidants, such as ethoxyquin. After a feed is made, the interaction between cationic minerals (such as iron, copper, zinc, magnesium, and manganese) catalyzes oxidation of vitamins and unsaturated fatty acids, which results in reduced nutritional quality. A well-made feed contains several additives and ingredients that serve to slow the oxidation of essential nutrients in a finished feed. Traditional feed formulation may include one or two of these feed protectants, but the production of complete feeds should incorporate a more thorough approach.

Manufacturing wholesome feeds should include the following steps to maintain the quality of the feed:

- Addition of antioxidants, e.g., ethoxyquin
- Ingredient arrival testing
- Stabilized Vitamin E supplementation
- Stabilized Vitamin C supplementation
- Extrusion pasteurization
- Mold inhibitors
- Mycotoxin binders
- Feed quality assurance testing

18.6 Practical Feed Formulation

Practical feed formulations generally contain no more than six to eight macro ingredients. The nutrient data matrix of each ingredient is utilized to meet minimum and maximum species nutritional restrictions. Nutrient restrictions are established based on experience, and generally meet and in some cases, exceed nutritional requirements. Data reports indicating the quantity of nutrients within each ingredient should be periodically updated, as seasonal changes in feedstuffs occur. In addition, a digestibility factor for each nutrient within the ingredient needs to be assigned based on species being fed. The quality of the matrix information greatly impacts the nutritional quality of the finished feed. The minimum and maximum species nutritional restrictions along with the nutrient database of each ingredient, the digestibility of the ingredient, and ingredient costs are then used in linear computer programs to optimize the mixture of ingredients that best meet the nutrients considered. The level of dietary energy is adjusted to provide the optimum protein to energy ratio to meet the nutritional requirements of the size and species being reared. The digestible amino acid profile of the protein is balanced for essential amino acids.

Even though both catfish and tilapia are omnivores, digestibility of feedstuffs is different between the species. Although it is sometimes done, it is not appropriate to feed a diet formulated to rear catfish in ponds to tilapia in recirculating systems. A currently recommended diet makeup for tilapia would follow the guidelines shown in Table 18.3. More information on digestibility of protein, fat, and carbohydrates is provided in Table 18.4 for Chinook salmon, rainbow trout, channel catfish, and blue tilapia (NRC, 1993). Table 18.5 provides true amino acid availability and protein digestibility values for certain feed ingredients for Atlantic salmon and channel catfish (NRC, 1993). Table 18.6 summarizes net absorption of phosphorus from various sources by channel catfish, common carp, and rainbow trout (NRC, 1993), and Table 18.7 provides specific information on digestibility coefficients of feedstuffs for tilapia *O. niloticus* (Lovell, 1989).

Wilson and Poe (1985) have shown that feed manufactured using extrusion as compared to pelleted processing increased the digestibility of energy but had no effect on the digestibility of protein. Popma (1982) described the difference in digestible energy between catfish and tilapia fed the same feedstuffs. Differences in digestible energy of key ingredients used in both catfish and tilapia commercial feeds are summarized in Table 18.8.

Table 18.3 Nutrient Composition of Typical Tilapia Feeds (Minimum or maximum expressed as g/100 g of finished feed.)

	Nutrient Composition of Typical Tilapia Feeds			
Fish Weight Nutrient	<2.0 Gram	2 to 10 Gram	10 to 50 Gram	50 to 545 Gram
Energy, digestible – Min	4.0 Kcal/g	3.8 Kcal/g	3.5 Kcal/g	2.9 Kcal/g
Protein – Min	48	45	40	36 or 32
Lipid – Min.	10	10	10	10 to 5
Fiber – Max.	4	4	5	5
Ash – Max.	7	7	9	9
Starch – Min.	12	14	20	24
Calcium – Max.	1	1	1	1.5
Avail. Phosphorus – Min.	0.6	0.6	0.6	0.6
Lysine – Min.	2	2	1.9	1.7
Methionine – Min.	0.9	0.85	0.85	0.7
Threonine – Min.	1.2	1.2	1.2	1

Ingredients selected for use in tilapia feeds can significantly impact the digestibility of the finished feed. Growth, feed conversion, and subsequently generated pollution are directly related to the degree of digestibility of the finished feed and the amount and type of feces produced. Feedstuff selection for recirculating systems is not only influenced by unit costs for energy, protein, amino acid composition and ingredient digestibility but also the level of phosphorus. The phosphorus/nitrogen (P/N) content of many ingredients such as animal by-product meals, with some exceptions such as herring, feather and blood meal (see Table 18.9) have a high P/N ratio. Generally, plant protein ingredients such as soybean and corn gluten meals have the desirable characteristic of a lower P/N ratio, which makes them appropriate for use in low environmental impact diet formulations.

Table 18.4 Apparent Digestibility of Protein, Fat, & Carbohydrate in Diet Ingredients for Chinook salmon, Rainbow Trout, Channel Catfish, and Blue Tilapia (NRC, 1993)

		Protein (%)				Lipid (%)			Carbohydrate (%)		
Ingredient	International Feed No.	Chinook Salmon	Rainbow Trout	Channel Catfish	Blue Tilapia	Rainbow Trout	Channel Catfish	Blue Tilapia	Rainbow Trout	Channel Catfish	Blue Tilapia
Alfalfa meal	1-00-023	-	61[a]	-	66[b]	71[c]	51[d]	-	-	12[d]	27[b]
Blood meal	5-00-381	30[c]	69[a]	74	-	-	-	-	-	-	-
Casein	5-01-162	-	95[a]	97[f]	-	-	-	-	-	-	-
Canola meal	5-06-145	79[c]	-	-	-	-	-	-	-	-	-
Corn grain	4-02-935	-	95[e]	60[d]	84[b]	-	76[d]	90[b]	-	66[d]	45[b]
cooked		-	-	66[d]	79[b]	-	96[d]	-	-	78[d]	72[b]
Corn gluten meal	5-28-242	-	87[a]	-	-	-	-	-	-	-	-
Cottonseed meal	5-01-621	-	76[a]	83[g]	-	-	88[d]	-	84[a]	17	-
Fish, anchovy meal	5-01-985	92[c]	-	-	-	-	-	-	-	-	-
Fish, herring meal	5-02-000	91[e]	87[a]	-	-	97[e]	-	-	-	-	-
Fish, manhaden meal	5-02-009	83[e]	-	88[g]	85[b]	-	97[d]	98[b]	-	-	-
Meat and bone meal	5-00-388	85[c]	82[d]	78[b]	-	77[c]					
Poultry by-product meal	5-03-798	74[c]	68[c]	-	-	-	-	-	-	-	-
Poultry, feathers, hydrolyzed	5-03-795	71[c]	58[c]	74[d]	-	68[c]	83[d]	-	-	-	-
Soybean meal, 44%	5-04-604	-	-	-	94[b]	-	-	-	-	-	54[b]
Soybean meal, 48%	5-04-612	77[c]	83[a]	93[g]	-	-	81[d]	-	-	-	-
Starch, corn (uncooked)											
50%		-	-	-	-	-	-	-	24[a]	-	55[f]
25%		-	-	-	-	-	-	-	-	-	61[f]
Starch, corn (cooked)											
50%		-	-	-	-	-	-	-	52[a]	-	66[f]
25%		-	-	-	-	-	-	-	-	-	78[f]
Wheat middlings	4-05-205	86[c]	76[a]	72[d]	-	-	-	-	-	-	-
Wheat grain	4-05-268	-	-	92[g]	90[b]	-	96[d]	85[b]	-	59[g]	61[b]
extruded, 44%		84[g]	-	-	-	-	-	-	-	-	-

NOTE: Dashes indicate data were not available. [a]Smith (1977) and Smith et al. (1980) determined by metabolism chamber, single ingredient fed.; [b]Popma (1982) determined by indicator method, feces collected by frequent removal from water, ingredient fed in mixed diet; [c]Cho et al. (1982) determined by indicator method, feces collected from a settling column outside the fish tank, ingredient fed in mixed diet; [d]Cruz (1975) determined by indicator method, feces collected by surgical excision, single ingredient fed; [d]Cruz (1975) determined by indicator method, feces collected from water, ingredient fed in mixed diet; [f]Saad (1989) determined by indicator method, feces collected by surgical excision, ingredient fed in mixed diet; [g]Wilson and Poe (1985) determined by indicator method, feces collected by surgical excision, ingredient fed in mixed diet.

Table 18.5 True Amino Acid Availability and Protein Digestibility Values for Certain Feed Ingredients for Atlantic Salmon and Channel Catfish (NRC, 1993)

Feed Ingredient Fish Species	International Feed No.	Protein (%)	ARG (%)	CYS (%)	HIS (%)	ILE (%)	LEU (%)	LYS (%)	MET (%)	PHE (%)	THR (%)	TRP (%)	TYR (%)	VAL (%)
Canola meal	5-06-145													
Atlantic salmon		91.4	96.7	97.1	95.0	87.3	85.0	92.0	99.9	89.2	93.2	-	92.9	83.8
Corn, grain	4-02-935													
Channel catfish		-	-	82.0	90.3	67.9	87.5	96.5	70.5	81.8	69.8	-	77.5	74.4
Corn, gluten meal	5-28-241													
Atlantic salmon		95.0	99.9	90.8	94.5	90.4	88.4	99.9	93.8	91.2	92.0	-	92.0	91.3
Cottonseed, meal	5-01-621													
Channel catfish		-	90.6	-	81.6	71.7	76.4	71.2	75.8	83.5	76.7	-	73.4	76.1
Fish, herring meal (flame dried)	5-02-000													
Atlantic salmon (steam dried)		93.8	95.3	86.2	93.8	91.9	94.1	92.3	87.6	92.4	93.2	92.9	95.4	91.4
Atlantic salmon (low temperature)		82.6	94.1	94.1	88.2	89.0	89.0	90.1	88.6	88.9	94.9	56.7	90.2	88.3
Atlantic salmon		88.8	93.8	95.6	92.6	94.7	94.1	95.8	92.0	93.4	99.8	86.2	96.5	93.5
Fish, menhaden meal	5-02-009													
Atlantic salmon		88.5	86.8	92.0	91.1	88.5	90.1	87.6	83.6	87.4	88.4	89.0	92.1	86.3
Channel catfish		-	91.0	-	84.5	87.1	89.0	86.4	83.1	87.3	87.4	-	88.8	87.1
Meat and bone meal	5-00-388													
Channel catfish		-	87.9	-	82.2	80.8	82.4	86.7	80.4	85.4	76.3	-	83.1	80.8
Peanut meal	5-03-650													
Channel catfish		-	97.7	-	89.4	93.3	95.1	94.1	91.2	96.0	93.4	-	94.5	93.3
Rice bran	4-03-928													
Channel catfish		-	94.2	-	83.4	87.5	90.5	94.7	88.2	89.5	88.2	-	93.7	89.2
Soybean meal	5-04-604													
Atlantic salmon		88.3	86.7	-	86.4	79.2	75.9	83.6	94.0	78.7	84.5	50.3	83.0	77.3
Channel catfish		-	96.8	-	87.9	79.7	83.5	94.1	84.6	84.2	82.2	-	83.3	78.5
Wheat middlings	4-05-205													
Channel catfish		-	95.1	-	94.5	87.8	89.9	96.3	82.8	93.0	89.1	-	89.1	90.1

Abbreviations: ARG = arginine, CYS = cysteine, HIS = histidine, ILE = isoleucine, LEU = leucine, LYS = lysine, MET = methionine & cysti (s) ne, PHE = phenylalanine & tyrosine, THR = threonine, TRP = tryptophan, TYR = tyrosine, and VAL = valine.

Table 18.6 Net Absorption of Phosphorus from Various Sources by Channel Catfish, Common Carp, And Rainbow Trout (NRC, 1993)

Source	International Feed Number	Channel Catfish (%)	Common Carp (%)	Rainbow Trout(%)
Animal Products				
Casein	5-01-162	90[a]	97[b]	90[b]
Egg albumin	–	–	71[b]	–
Anchovy fishmeals	5-01-985	–	–	–
Brown fishmeals	–	–	24[b]	74[b]
Menhaden fishmeals	5-02-009	60[c]	–	–
White fishmeals	–	–	0–18[b]	66[b]
Inorganic phosphates				
Calcium, monobasic	6-01-082	94[c]	94[b]	94[b]
Calcium, dibasic	6-01-80	65[c]	46[b]	71[b]
Calcium, tribasic	6-01-084	–	13[b]	64[b]
Potassium, monobasic	–	–	94[b]	98[b]
Sodium, monobasic	6-04-288	90[c]	94[b]	98[b]
Plant products				
Corn, ground	4-26-023	25[c]	–	–
Phytate	–	1[c]	8–38[b]	–
Rice bran	4-03-928	–	25[b]	19[b]
Soybean meal, dehulled	5-04-612	20[a]	–	–
Wheat germ	5-05-218	–	57[b]	58[b]
Wheat middlings	4-05-205	28[c]	–	–
Yeast, brewers	7-05-527	–	93[b]	–

[a]Data from Wilson, R. P., E. H. Robinson, D. M. Gatlin, III, and W.E. Poe, 1982. Dietary phosphorus requirement of channel catfish. J. Nutr. 112:1197–1202. Values are expressed as percent apparent absorption.

[b]Data from Ogino, C., T. Takeuchi, H. Takeda and T. Watanabe, 1979. Availability of dietary phosphorus in carp and rainbow trout. Bull. Jpn. Soc. Sci. Fish. 45:1527–1532.

[c]Data from Lovell, R. T., 1978. Dietary phosphorus requirement of channel catfish (*Ictalurus punctatus*). Trans. Am. Fish. Soc. 107:617–621. Values are expressed as percent apparent absorption.

Table 18.7 Digestibility Coefficient of Feedstuffs for Tilapia (*O. niloticus*) (Lovell, 1989a)

Feedstuff	Percentage Digestibility			
	Protein	Fat	Carbohydrate	Gross energy
Fish meal	**84.8**	**97.8**	–	**87.4**
Meat and bone meal (high grade)	77.7	–	–	68.7
Soybean meal	94.4	–	53.5	72.5
Corn (uncooked, mixed with fish meal)	–	–	65.4	–
Corn (cooked)	78.6	–	72.2	67.8
Wheat	89.6	84.9	60.8	65.3
Wheat bran	70.7	–	–	–
Alfalfa meal	65.7	–	27.7	22.9
Coffee pulp	29.2	–	–	11.4

Table 18.8 Differences in Digestibility of Energy in Feedstuffs by Catfish and Tilapia (*O. niloticus*) (Lovell, 1989)

	Ingredient Digestible Energy (Mcal/g)	
	Catfish	Tilapia
Alfalfa, 17% Protein	0.67	1.01
Corn Grain		
Raw	1.10	2.46
Processed	2.53	3.02
Menhaden Fish Meal	3.90	4.04
Molasses	3.47	2.94
Soybean Meal, 48% Protein	2.58	3.34
Wheat Flour	2.55	2.89

Table 18.9 Phosphorus to Nitrogen Ratios for a Variety of Feed Ingredients

Ingredient	Protein	Nitrogen	Phosphorus	P/N
Herring Meal	72	11.52	1.00	0.087
Feather Meal	85	13.60	0.70	0.051
Corn Gluten	60	9.60	0.70	0.073
Peanut Meal	47	7.52	0.60	0.080
Soybean Meal	48	7.68	0.65	0.085
Wheat, Soft	11	1.73	0.30	0.174
Yellow Wheat	9	1.42	0.25	0.176
Poultry Meal	58	9.28	2.40	0.259
Menhaden Meal	62	9.92	3.00	0.302
Wheat Middlings	17	2.72	0.91	0.335
Meat/Bone Meal	50	8.00	4.70	0.588
Blood Meal	80	12.80	0.22	0.017

The P/N ratio is an example of derivative criteria, which can be added to a matrix presentation of nutrient specifications. Cho et al. (1994) has recorded the P/N ratio of several ingredients, and this information can be added to the nutrient specification matrix. The nutrient matrix will be discussed later, but it is important to know that actual nutrient levels as well as derivatives such as the P/N ratio can be used to assist in formulating low impact aqua feeds. See Table 18.9 for a summary of P/N ratios for a variety of feedstuffs.

Several states (USA) have established discharge standards to regulate the amounts of phosphorus that may be discharged by fish farms and hatcheries. Properly designed recirculating systems can easily meet these standards by simply discharging 1% or less of the rearing capacity water on a daily basis (see Chapter 6 for more details on managing waste). However, discharge standards for small volume releases are continually under review, and regulations will steadily become more restrictive. The consumer generally wishes to be environmentally sensitive, so marketing efforts for aquaculture produced fish incorporate references to the clean and wholesome image of an environmentally friendly production methodology. This, in turn, can and does result in increased values for aquaculture fish in the open market.

18.7 Important Aspects Of Aquaculture Feeds

Fish growth and performance can be quickly compromised by poorly digested feed that negatively impacts water quality. The type of feed influences fecal consistency. Dissolved solids from feed affect the color of the culture system water and this indirectly affects fish behavior, sometimes beneficially. The type of pellet and the process used to manufacture the pellet (steam, extrusion, expanded) will have an impact on the water quality as well as on the digestion efficiency of the fish. This is reviewed later in more detail. Designing feeds for low environmental impact is particularly critical when applied to RAS.

Standard nutrient requirements (NRC, 1993) cannot normally be applied to practical feed formulations. These requirements were established for small rapidly growing fish in ideal environments, and are not adequate for high-density commercial rearing situations. Therefore, nutritional requirements must be specifically established for each enterprise, based on the species to be grown and the type of aquaculture system used. Commercial feed formulations are intended to meet or exceed all nutritional requirements with quality products available at reasonable prices, so it is the responsibility of the aquaculturist to know what the species requires and to purchase feed appropriately. While the water column itself contains trace amounts of nutrients independent of the feed that is introduced, these nutritional contributions of vitamins, minerals, key lipids, protein and energy from the environment are negligible and cannot be counted on to contribute to growth.

While many feed ingredients can be used to meet nutritional requirements, not all ingredients are suitable for feeds used in RAS systems. Many are not sufficiently digestible. Some ingredients contribute to the production of excessive solids, increase visceral fat deposits, create high levels of iron, inhibit the absorption of zinc, and can result in undesirable yellowing of fillets.

Ingredient Quality

Quality feeds can be made only with quality ingredients. Quality assurance begins at the feed mill with ingredient reception, as incoming products are inspected and tested to assure that certain molds or histamines are not present. If ingredients contaminated with these molds are used in the feed, the animals consuming the feed could develop problems. Mycotoxin is a particularly dangerous contaminant that can be found in fish food if affected ingredient lots are used to make the feed.

One of the major ingredients in fish food is fishmeal. This ingredient is tested to determine the levels of histamine in the fishmeal, and the lot

will be rejected if levels are beyond acceptable limits. Histamine is formed by bacterial decarboxylation of the amino acid histidine. Histamine is frequently found in brown fish meals (from sardines and mackerel). Feeding diets rich in histamine to trout causes discoloration of skin and erosion of the stomach.

Mycotoxins include various toxins produced by fungi, i.e., mushrooms, molds, or yeasts. Examples important to fish include aflatoxins, fumonisin and vomitoxin. Aflatoxins are produced by *Aspergillus* and fumonisin and vomitoxin by *Fusarium* molds. Aflatoxins are among the most carcinogenic substances known. *Aspergillus* is in soil and can infect cotton, various grains, corn and peanuts undergoing microbiological deterioration whenever conditions are favorable for its growth. Favorable conditions include high moisture content and high temperature. Vomitoxin is deoxynivalenol (DON).

Coldwater species, such as trout, are more sensitive to aflatoxin than warmwater species. Corn is rejected if aflatoxin levels exceed 20 ppb or if fumonisin[2] levels are found to exceed 1 ppm. Wheat is tested to determine the levels of deoxynivalenol (DON) is rejected if levels exceed 5 ppm. DON originating with wheat reduces growth and growth rates, increases morbidity, and promotes tumors in salmon livers (P.D. Maugle).

The standards for mycotoxin tolerance were established for terrestrial animals, not for fish. Fish are more sensitive to mycotoxin than the federal standards would indicate. Therefore, do not rely on the federal standards to gauge the safety of fish food. The feed

[2] Fumonisins are environmental toxins produced by the molds *Fusarium moniliforme* (*F. verticillioides*), *F. proliferatum*, and other *Fusarium* species that grow on agricultural commodities in the field or during storage. These mycotoxins have been found as contaminants, mainly in corn, worldwide. More than ten types of fumonisins have been isolated and characterized. Of these, fumonisin B_1 (FB_1), fumonisin B_2 (FB_2), and fumonisin B_3 (FB_3) are the major fumonisins produced in nature. The most prevalent of these mycotoxins in contaminated corn is FB_1, which is believed to be the most toxic.
Referenced from:
1) Thiel, P.G., Marasas, W.F.O., Sydenham, E.W., Shephard, G.S. and Gelderblom, W.C.A., 1992. The implications of naturally occurring levels of fumonisins in corn for human and animal health, Mycopathologia 117:3–9.
2) Musser, S.M. and Plattner, R.D., 1997. Fumonisin composition in cultures of *Fusarium moniliforme*, *Fusarium proliferatum*, and *Fusarium nygami*. J. Agric. Food Chem. 45:1169–1173.
3) Ashley, L. M., 1970. A symposium on diseases of fishes and shellfishes. Am. Fish. Soc. Spec. Pub. 5:366–379.

manufacturers are aware of this, so they generally incorporate one or more specific mycotoxin binders into the feed, such as bentonite, zeolite, yeast, ball clay, and other additives. However, no specific data on the efficacy of these additives in fish food is available.

In addition to the lack of specific standards for fish and the unquantified results of the mycotoxin mitigating additives, the quality of the final nutritional content of feed will vary based on the natural differences of the ingredients from seasonal variations and location of origin. Therefore, the aquaculturist must continually assess the performance of the nutritional matrix by carefully monitoring fish health and growth rates. The quality of the nutrient matrix in feed formulation programs is indispensable. Seasonal nutrient variability within key ingredients can impact extrusion characteristics, and create variability in final feed characteristics.

Fishmeal is used to meet essential amino acid requirements as well as maintain overall palatability of the finished feed. Fishmeals are sold by protein levels, ash levels, and the temperature of processing. Principal indicators of fishmeal quality are that it be Salmonella negative, contain less than 500 ppm histamine and be stabilized during manufacture with at least 250 ppm antioxidant, ethoyquin (Santoquin). Lower levels of histamine are preferable, particularly in starter feeds.

OXIDATION OF DIETARY FATS AND VITAMINS

Unsaturated fatty acids in feeds, fish oil, and other ingredients containing them, are likely to undergo oxidation, which, practically speaking, cannot be completely prevented, but only retarded. Fish feeds are particularly rich in unsaturated fats because of their peculiar nutritional needs. Therefore fish feeds must be properly stored and protected by the addition of antioxidants such as ethoxyquin (Santoquin), BHA (butylated hydroxyanisole, BHT (butylated hydroxytoulene), and others. Ethoxyquin is highly effective, followed closely by BHT and BHA. Proper storage requires keeping feed in a cool dry area, generally no longer than 3 months. When feeds react with oxygen, unsaturated fatty acids are destroyed, and chemicals, called peroxides, are produced which destroy other nutrients in feed. Among those most vital and readily destroyed are various vitamins. Unprotected forms of vitamins E and C are especially subject to destruction. Old feeds or feeds improperly stored or unprotected are a common cause of nutritional problems in fish, especially young fish. Oxidation of fish feeds or ingredients may be assessed by commercial testing laboratories by measuring thiobarbituric acid may be assessed by commercial testing laboratories by measuring thiobarbituric acid (TBA) or peroxide value (PV). Feed having a TBA

value of about 8 mg/kg or greater and a PV value around 8meq/kg oil or greater should be considered unacceptably damaged by oxidation (Ketola et al., 1989).

FORMULATION

The organic materials that provide energy to the fish are either proteins, carbohydrates, or lipids. Vitamins and minerals are also necessary dietary ingredients.

PROTEINS

Proteins are composed of amino acid chains of varying proportions; most proteins contain approximately 22 amino acids. Ten of these amino acids are deemed to be "essential" and cannot be synthesized by fish, therefore, they must be added to the diet. Two other amino acids, cystine and tyrosine, are conditionally required because they can be synthesized from two other essential amino acids if supplied in sufficient quantities. Cystine can be synthesized (by way of cysteine) from the essential amino acid methionine. Tyrosine can be synthesized from the essential amino acid phenylalanine. These transformations are irreversible and represent only a part of the functions for methionine and phenylalanine; therefore these conditionally required amino acids (cystine and tyrosine) can spare part of the quantitative requirements for methionine and phenylalanine.

Fishmeal, which generally contains all the essential amino acids, is a common protein source. Supplements can be added to feed containing alternate proteins as necessary to meet the minimum requirements. Proteins comprise a significant portion of the salmonid diet, generally ranging between 35 and 55%, depending on the age of the fish, with smaller fish having a higher metabolic rate, thereby requiring the higher protein levels.

Fishmeal is used to meet essential amino acid requirements as well as maintain overall palatability of the finished feed. Commercially available fishmeals are graded and sold by protein levels, ash levels, and the temperature of processing.

LIPIDS

Lipids are the most efficient source of energy for fish, but do not entirely replace the other two sources. Nutritionally important lipids include triglycerides and phospholipids. Triglycerides include fats and oils. Lipids are categorized into three groups: fats, oils, and waxes. Fats and oils are the primary energy source in feed and are important for a

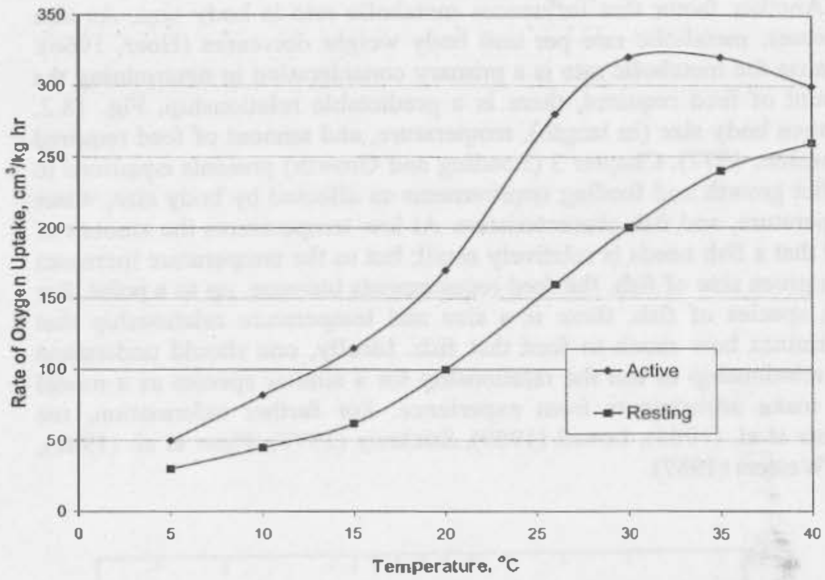
balanced ration. Fats and oils are nearly the same chemically and nutritionally, with the main difference being the melting point. Three phospholipids are lecithin, cephalin, and sphingomyelin. The available energy content of lecithin was found to be 6.5 kcal/g for simple-stomached animals. This represents the maximum theoretical energy value of lecithin, since approximately 25% of the molecule is phosphoric acid and choline. Little work has been done on the energy value of cephalins and sphingomyelins. These compounds, however, are not found in significant amounts in foods or feeds. Trout feed typically contains between 8% to 15% fat content. Some farmers advise feeding lower fat feed before transport, as fish fed a low fat ration fare better.

CARBOHYDRATES

Carbohydrates are not normally included as a large part of the salmonid diet due to their low nutritional content and poor digestibility of energy. Although they are an inexpensive source of energy, they do not supply any essential nutrients that cannot be obtained elsewhere in the diet. Additionally, excess carbohydrates can cause liver problems leading to death in salmonids. Trout diets should contain no more than 12–20% of maximum digestible carbohydrates. However, carbohydrates are important components of the feed for other reasons. In the form of starches, carbohydrates play an important role in the binding of the feed. Cooking, extrusion, and expansion are methods used to improve digestibility and binding capabilities of the salmonid pellets.

18.8 PHYSIOLOGICAL RELATIONSHIPS

Fish are poikilothermic (cold-blooded) animals. Their body temperature and metabolic rate are largely determined by water temperature. Species responses differ somewhat, but the concepts are similar. The goldfish (*Carassius auratus*) provides a typical example, Fig. 18.1, of the relationship between metabolic rate and temperature (Fry and Hart, 1948). The top bottom graph in Fig. 18.1 represents the difference between active and standard (resting) metabolism. Metabolic rate increases as temperature increases, up to a point, beyond which there is a plateau. At even higher temperatures, their metabolic rate decreases, and eventually the fish die.



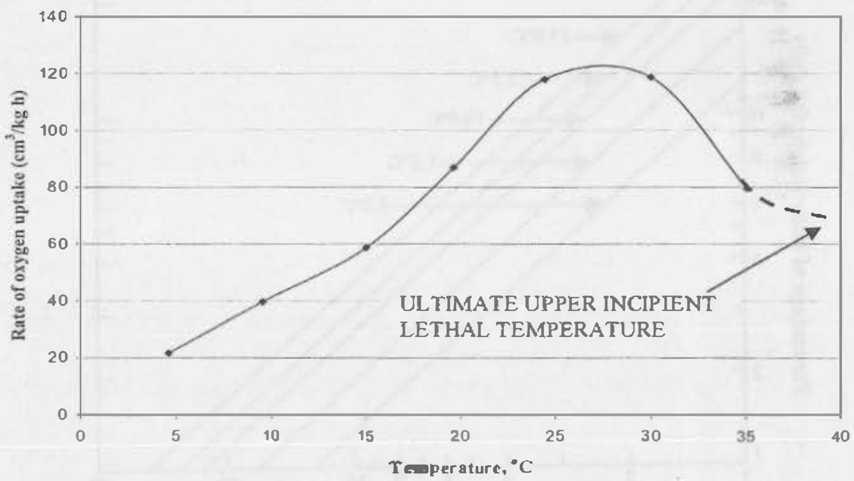


Figure 18.1 Metabolic rate as measured by oxygen consumption in the Goldfish (Fry And Hart, 1948; Taken from Wheaton, 1977, pg. 106).

Another factor that influences metabolic rate is body size. As size increases, metabolic rate per unit body weight decreases (Hoar, 1966). Because the metabolic rate is a primary consideration in determining the amount of feed required, there is a predictable relationship, Fig. 18.2, between body size (in length), temperature, and amount of feed required (Wheaton, 1977). Chapter 3 (Loading and Growth) presents equations to predict growth and feeding requirements as affected by body size, water temperature, and fish characteristics. At low temperatures the amount of feed that a fish needs is relatively small; but as the temperature increases for a given size of fish, the feed requirements increase, up to a point. For each species of fish, there is a size and temperature relationship that determines how much to feed that fish. Ideally, one should understand this relationship or use the relationship for a similar species as a model and make adjustments from experience. For further information, see Klontz et al. (1985), Lovell (1989), Stickney (1979), Piper et al. (1982), and Westers (1987).

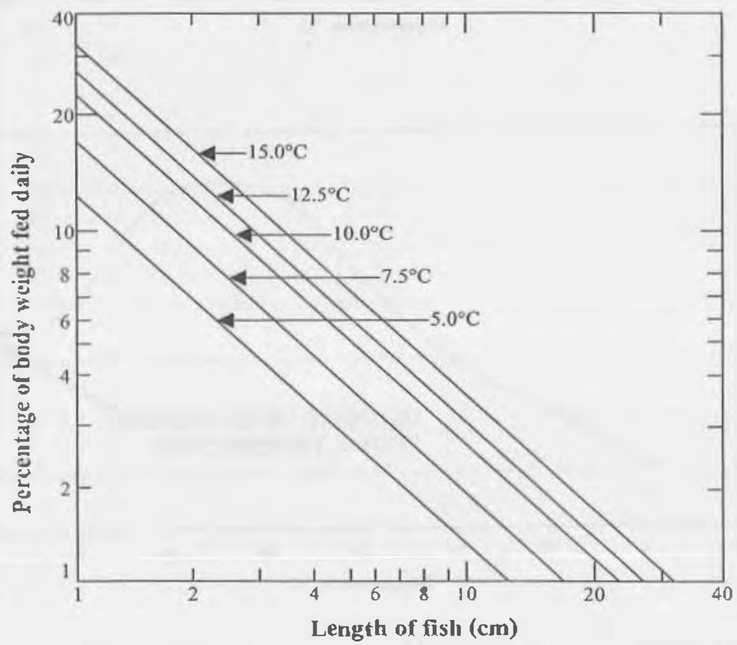


Figure 18.2 Feed required by Trout as related to temperature (C) and body size (from Wheaton (1977) Fig. 13.62 pg. 577).

18.9 Water Chemistry and Dietary Needs

Water chemistry influences physiology and nutrition of fish. For example, rainbow trout (*Oncorhynchus mykiss*) living in seawater experience osmotic pressure that causes water to be continually lost from the body fluids which poses a danger of dehydration. To counteract osmosis and avoid dehydration, the trout drinks saltwater and eliminates excess salts by active transport across the gills and by urinary excretion. The opposite is true for trout in fresh water. Water moves osmotically into the tissues of the fish, through the skin, gills, etc. Therefore, freshwater fish do not drink but rather excrete copious amounts of dilute urine. This difference in physiological behavior significantly affects nutritional requirements. The drinking of seawater contributes noticeably to nutrient intake as can be seen by examination of data by Shehadeh and Gordon (1969), Table 18.10. In fresh water, there was no intake of minerals by drinking. When rainbow trout were reared in a 30% mixture of sea water to fresh water (30/70), there was a marked increase in drinking (42 mL/kg of body weight per day); and at full strength (100%) sea water there was an even greater rate of drinking (129 mL/kg of body weight per day).

Table 18.10 compares estimates of mineral intakes by feed to intakes by drinking seawater. Estimates of mineral intakes by feed assume a hatchery diet fed to trout at a normal feeding rate of 1% of body weight/day. These estimates show that intake of calcium by drinking represented about one-half the amount consumed in feed, Table 18.10. The amounts of magnesium and sodium consumed by drinking seawater were about 10 to 13 times higher than by feed. Intake of chloride was much higher than obtained from feed. Absorption of these minerals from drinking was considerable, Table 18.10. Calcium absorption was moderate, 3–17 mg. The amounts of magnesium, sodium, and chloride absorbed from water were much higher than obtained from feed. This shows that when trout are held in seawater, drinking contributes substantially to the nutrient intake.

The gills also contribute to the absorption of calcium and other divalent metal ions even in fresh water. A study of brook trout (*Salvelinus fontinalis*) by McCay et al. (1936) involved three replicates of 50 fish. Twenty-five trout from each group were analyzed for calcium at the start and the rest of the trout were fed a diet of liver for 12 weeks. Liver contains very little calcium. Chemical analyses of carcasses at the beginning and at the end of the study, Table 18.11, showed that feed provided only about 1.1 milligrams of calcium per fish, whereas the total calcium deposited in the carcass was much more. Computations showed

that the feed could have only provided about 19% of the calcium accumulated. Since these fish do not drink in fresh water, the rest of the calcium was likely obtained by direct absorption through the gills and skin. The gills constantly process water, and there is a significant uptake of divalent metal ions by the gills in the fresh water (Lovelace and Podoliak, 1952; Reid et al. 1959; Boroughs et al. 1957; Smith, 1930).

Table 18.10 Drinking Seawater Contributes to Nutrition of Rainbow Trout

Nutrient Intakes per kg of Body Weight/Day Water (fresh, sea or mixture**)		Fresh water (100%)	SW/FW (30/70)	SW/FW (50/50)	Sea water (100%)	Nutrient Intake from Feed ***
Intake by Drinking*						
Fluid*	(mL)	0	42	95	129	1
Mineral intake (computed)****						
Calcium	(mg)	0	5	19	52	100
Magnesium	(mg)	0	19	64	174	15
Sodium	(mg)	0	147	499	1355	100
Chloride	(mg)	0	266	902	2451	100
Adsorption of minerals*						
Calcium	(mg)	-	3	4	17	-
Magnesium	(mg)	-	1	9	74	-
Sodium	(mg)	-	92	437	1288	-
Chloride	(mg)	-	195	845	2400	-

*Data from Shehadeh and Gordon (1969).

** FW = fresh water; SW = sea water.

*** Estimated nutrient intake by way of feed containing 1% calcium, sodium, and chloride, and 0.15% magnesium. Feed intake = 1% of bw/day.

**** Mineral intakes computed from fluid data and concentrations of minerals in mixtures of 30%, 50% and 100% sea water. Concentrations for 100% sea water were: calcium, 400 mg/Liter; magnesium, 1.350 mg/Liter; sodium, 10,500 mg/Liter; and chloride, 19,000 mg/Liter (Spotte, 1970).

Table 18.11 Uptake by Brook Trout of Calcium (Ca) from Food and Water (43 mg/L Ca) (McCay et al. 1936)

Measurement		Start	End	Gain
Body weight	(g/fish)	3.70	4.43	0.73
Carcass Ca	(mg/fish)	14.2	20.0	5.8
Feed fed	(g/fish)		18.7	
Feed Ca fed	(mg/fish)		1.1	

Percentage of Ca deposited (an indication of uptake)
from: $\frac{1.1\ (100)}{5.8} = 19\%$
From water (calculated as remainder): 100–19 = 81%

18.10 Functional Anatomy of Digestion

The gastrointestinal tract influences the fish's nutritional needs. The intestinal tract of a 2 1/2-week-old embryo of a brown trout (*Salmo trutta*), developing at 12°C, is basically a straight tube, very simple, with the yolk sac attached to provide nutrition as shown in Fig. 18.3 (Burnstock, 1959).

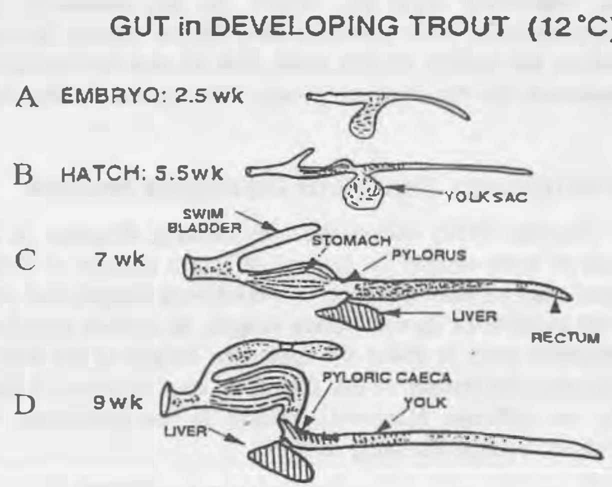


Figure 18.3 Gut development in Trout (12°C) from early state of its development at A) 2.5 weeks, B) 5.5 weeks, C) 7 weeks, and D) 9 weeks. (from Burnstock, 1959).

Brown trout embryos hatch in about 5 ½ weeks, at which time the stomach is only partially developed. At this stage of development, nutrition is provided by the yolk sac. At seven weeks, the intestinal tract is still not fully capable of processing food. At nine weeks, the fish swim up and begin to feed for the first time when their pyloric caeca are still rudimentary. There is still yolk material remaining that provides part of their nutrition. Because of under-development of the digestive tract, exogenous food provided for first-feeding should be highly digestible and contain high levels of energy and nutrients. Different species develop at different rates. Some species begin to feed in less than one week after hatching.

It is important to provide high quality nutrition to first-feeding fish at the appropriate time. If first-feeding fish do not receive adequate nutrition, the rest of that fish's life may be negatively impacted. There are a number of species of fish that are very difficult to feed initially. Studies with Atlantic salmon (*Salmo salar*) show a negative effect of oxidized dietary lipid on first feeding of these fry (Ketola et al. 1989). These salmon are most susceptible to oxidized lipids during the first few week of feeding, but with age they develop greater capabilities of handling oxidized lipids. There may be other fishes similar in this regard. Carnivorous fish, especially their fry, which do not ordinarily eat, oxidized lipids, appear to be most sensitive to oxidized dietary lipids in their diets. Therefore, the quality of fish meal, fish oil and fat-containing ingredients is important for the diets of young fish, especially the first-feeding fish.

Variations in Intestinal Tracts of Different Species

Figure 18.4 (Halver, 1959) shows the comparative diagram of gut length as a percent of body length for several different species of fishes. The gastrointestinal tract of rainbow trout is a relatively simple and short system, roughly 60 to 80% of its total body length. In catfish (*Ictalurus punactus*), the digestive tract is about 1.5 times the length of the fish. In carp (*Cyprinus carpio*) the length of the digestive tract is about 3 times the body length; in milkfish (*Chanos*), which is an herbivore, the digestive tract is about 5 times the body length.

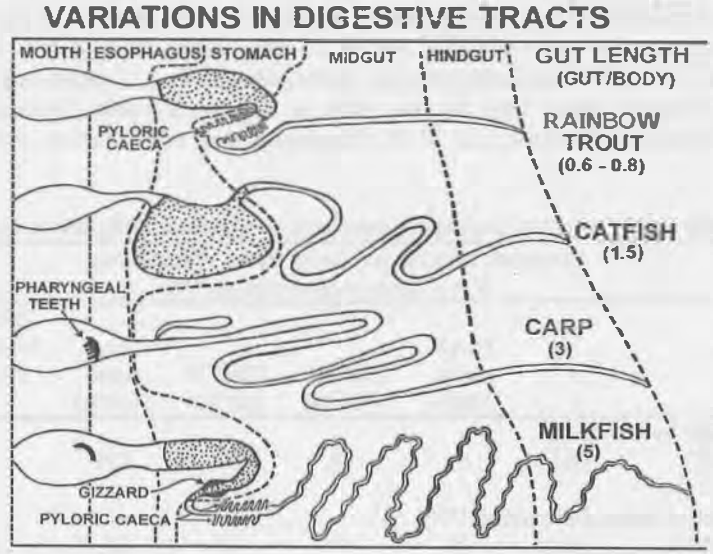


Figure 18.4 Variation in digestive tracts. Stippling indicates an acid secreting stomach. Rainbow trout (carnivore). Catfish (omnivore emphasizing animal sources of food, pouched stomach). Carp (omnivore emphasizing plant sources of food, pouched stomach). Milkfish (microphagous planktivore, tubular stomach with muscular gizzard).

There are other important differences in digestive capabilities between these fishes. The trout and the catfish both have acid producing stomachs; the carp, which is a member of the minnow family, does not have an acid producing stomach. Minnows in general do not have true stomachs and cannot produce hydrochloric acid. Acid affects their nutritional responses and needs. Later, we will see how the lack of acid in carp affects their ability to digest phosphorus, Table 18.12. The milkfish has a gizzard, apparently to grind its food. These fish have very long gastrointestinal tracts. Milkfish and carp tend to graze continuously, whereas carnivorous fish tend to eat fewer meals less frequently and retain food in their digestive tracts for longer periods.

Table 18.12 Comparative Availability of Phosphorus for Carp, Catfish and Trout (NRC/NAS, 1983)

Source of P	Solubility (g/100 cc)	Availability Carp	Catfish	Trout
$NaH_2PO_4 \cdot H_2O$	60	94	90	98
KH_2PO_4	33	94	–	98
$Ca(H_2PO_4)_2 \cdot H_2O$ (mono)	1.8	94	94	94
$CaHPO_4 \cdot 2H_2O$ (di)	0.03	46	65	71
$Ca_3(PO_4)_2$ (tri)	0.002	13	–	64
Fish meals	–	18–24	40	66–74
Casein	–	97	30	90
Yeast	–	93	–	91
Rice bran	–	25	–	19
Wheat germ	–	37	–	37
Phytate	–	3–18	Nil	0–19

18.11 Minerals

Phosphorus is an essential nutrient for fish. Figs. 18.5 and 18.6 show the results of an experiment in which Atlantic salmon (*Salmo salar*) were fed a phosphorus-deficient diet supplemented with different forms of phosphorus— either phytate phosphorus, meat and bone meal phosphorus, or a highly-soluble sodium dihydrogen monohydrate phosphate ($NaH_2PO_4 \cdot H_2O$) (Ketola, H.G., unpublished). Plant phosphorus in general is about two-thirds phytate phosphorus. Figures 18.5 and 18.6 show that phytate phosphorus supplements did not benefit salmon, whereas supplements of phosphorus in meat and bone meal and sodium phosphate significantly improve both growth and bone ash measurements. The same is true of trout and probably other fishes as well (NRC, 1983).

Table 16.12 summarizes reports reviewed by the National Research Council (1983) that compare the availability of various sources of phosphorus to common carp, channel catfish, and rainbow trout. For trout, highly soluble sodium phosphate ($NaH_2PO_4 \cdot H_2O$) and potassium phosphate (KH_2PO_4) are both highly available (98%), and monocalcium phosphate ($Ca(H_2PO_4)_2 \cdot 2H_2O$) is also readily available (94%). However, dicalcium phosphate ($CaHPO_4 \cdot 2H_2O$) and tricalcium phosphate ($Ca_3(PO_4)_2$) are less available (71 and 64%), following the trend of decreasing solubility. In carp, this decrease in availability is much more dramatic due to the lack of hydrochloric acid production necessary to solubilize di- and tri-calcium phosphate. Trout and catfish, both of which produce acid, were better able to solubilize and digest these phosphates.

These differences in nutritional capabilities relate to basic differences in physiological characteristics of these fishes.

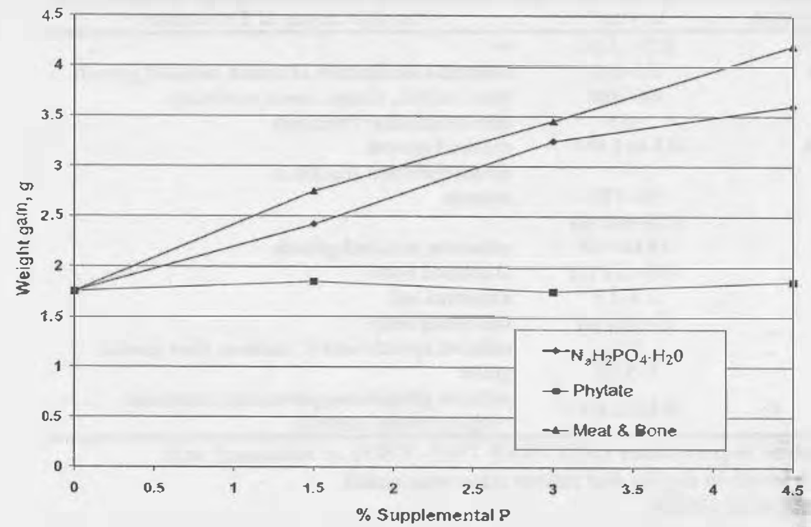


Figure 18.5 Availability of phosphorus to Atlantic Salmon and effect on growth.

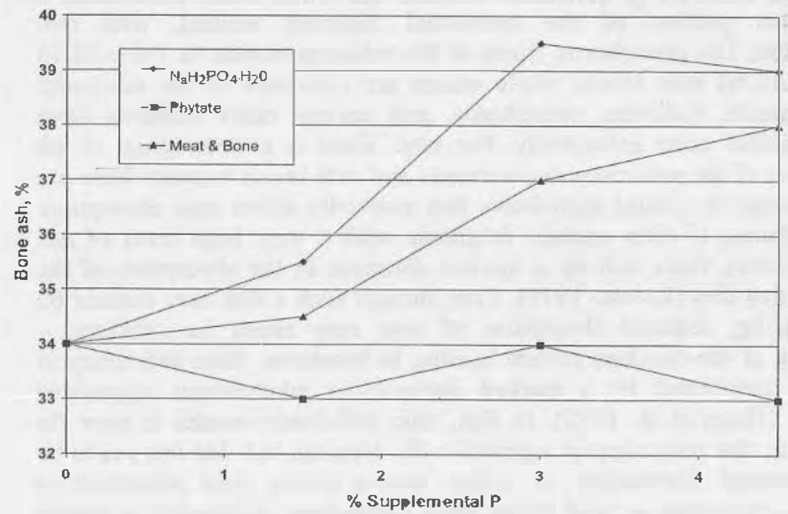


Figure 18.6 Availability of phosphorus (P) to Atlantic Salmon and effect on bone ash.

Table 18.13 General Summary of the Mineral Requirements of Various Fishes

Mineral	Levels*	Possible Signs of Deficiency
Ca%	0.1 - .3 (s)	—
P %	0.5–0.8	reduced calcification of bones, reduced growth
Mg	400–800	renal calculi, sluggishness, mortality
Na	ND	osmoregulation functions
K %	0.5 to 1.0**	reduced growth
Cl	—	osmoregulation functions
Fe	30–170	anemia
	150–400 (s)	
Zn	15 to>60	cataracts, reduced growth
	100–150 (s)	shortened body
Mn	2.4–13	abnormal tail
	10–100 (s)	shortened body
Cu	3–5	reduced cytochrome C oxidase, slow growth
I	1–5 (s)	goiter
Se	0.15 to 0.4	reduced glutathione peroxidase, muscular degeneration, anemia

*Tentative requirements (NRC/NAS 1983, 1993) or estimated safe (s) levels in mg/kg diet unless otherwise noted.
**Austic et al. (1989)

Table 18.13 shows a general summary of the mineral requirements of various fishes. The minerals requirements that must be met by feed are relatively difficult to determine because the water often contributes a significant portion of the nutritional minerals needed, with few exceptions, i.e., phosphorus. Some of the values presented in Table 18.13 are estimated safe levels, while others are estimates of the minimum requirements. Calcium, phosphorus, and several other minerals have been studied quite extensively. For zinc, there is a wide range in the estimates of the minimum requirements and safe levels because there are components in natural ingredients that markedly affect zinc absorption. For instance, if diets contain fishmeals with a very high level of ash (about 20%), there will be a marked decrease in the absorption of the zinc in that diet (Ketola, 1979). Even though such a diet may contain 60 mg zinc/kg, reduced absorption of zinc may result in cataracts, a clouding of the eye lens protein leading to blindness. Zinc deficiency in rats is manifested by a marked increase in inter-animal aggressive activity (Halas et al. 1975). In fish, zinc deficiency results in poor fin condition that may suggest aggressive fin nipping, but that has yet to be demonstrated. Deficiency of iodine causes goiter, first identified as thyroid carcinoma in wild brook trout (*Salvelinus fontinalis*) in waters low in iodine (Marine and Lenhart, 1911). Addition of iodine to the water reversed the condition. For further information on mineral

deficiencies in fish, see Halver (1989), Lovell (1989), and NRC/NAS (1981 and 1983).

Table 18.14 Fish Vitamin Requirements and Recommended Levels of Concentration in Diet

Vitamin	Level* (IU or mg/kg)	Signs (partial list)
A, IU/kg	2,500–5,000	retinal degeneration exophthalmia, corneal edema
D, IU/kg	500–2,400	poor growth fatty liver, tetany, droopy tail
E, IU/kg	25–100	anemia muscular degeneration
K, mg	1	prolonged clotting
C (Ascorbic acid), mg	50–100	scoliosis, lordosis hemorrhages, dark coloration of skin
Choline, mg	400–1,500	slow growth fatty liver

*Tentative minimum requirements as noted are expressed in IUs (International Units of activity) or mg/kg finished feed (NRC 1983, 1993). This table includes no margin of safety to compensate for losses due to pelleting, oxidation during storage or losses by leaching in water. Practical formulations must include appropriate overages to compensate for such losses. Stabilized forms of vitamins should be used especially for vitamins C and E and others (vitamins A, K and D).

18.12 Vitamins

High-density rearing conditions make it necessary for the commercial feed manufacturer to provide a complete and in some cases over formulate vitamins in their feed. The commercial feed manufacturer must provide a guarantee that the feed will retain the stated nutritional values for at least 3 months after production. Tables 18.14 and 16.15 summarize the requirements of fishes for vitamins and identify typical signs of deficiency. Vitamin C deficiency causes scoliosis and lordosis, which are deformities in the backbone that are not reversible. Early signs of vitamin C deficiency include hemorrhages in fins, skin, and internal organs. Halver (1989) specifically itemized the vitamin requirements for trout. Vitamin requirements for tilapia have been determined for only vitamin C (Stickney et al. 1984; Soliman et al. 1986), vitamin E (Satoh et al. 1987), and riboflavin and pantothenic acid (N.R.C., 1993). For further details on various forms of vitamin deficiency, see reports by NRC/NAS

(1981, 1983), Halver (1989), Lovell (1989), Poston (1986), Piper (1982), Stickney and Lovell (1977), Hilton and Slinger (1981), and Ketola (1976).

Table 18.15 Requirements of Fishes for Vitamins and Some Typical Signs of Deficiency

Vitamin	Level* (mg/kg)	Signs (Partial List)
Thiamin (B_1)	1–15	anorexia, convulsions
Riboflavin (B_2)	3–25	anorexia, dark coloration, incardination, cataracts, high mortality
Pyridoxine (B_6)	3–20	epileptic-like seizures, spiral swimming, rapid breathing
Pantothenate	10–50 100–200 (s)	clubbed gills sluggish
Niacin	10–28 150–200 (s)	anorexia, sunburn, ascites
Biotin	0.1–1.5	anorexia, anemia, mortality
Folic acid	1–2 5–15 (s)	anemia
Cyanocobalamin (B_{12})	0.015–0.05	anemia (?)

*Tentative minimum requirements are expressed in mg/kg finished feed (NRC 1983, 1993). This table includes no margin of safety to compensate for losses due to pelleting, oxidation during storage or losses by leaching in water. Practical formulations must include appropriate overages to compensate for such losses.

Stability of Vitamins in Feeds

(The following discussions, figures and tables until the Section on Vitamin Requirements and Stability – were extracted from a report by BASF, 1992)

Vitamin C (ascorbic acid) deficiency is one of the most common problems encountered in feeding fish, because vitamin C is normally very unstable during manufacture and storage of the diet (see Tables 18.16a and b). Losses of crystalline ascorbic acid in the diet may be great, especially during the pelleting process. For this reason, assuring an adequate delivery of vitamin C represents one of the major concerns in feeding fish. Table 18.16 shows the average stability of vitamins in feed stored at room temperature. Of particular importance is the rapid reduction of the natural forms of vitamin C (crystalline ascorbic acid) and vitamin E (alcohol form) when incorporated in feed. Therefore, most feed manufacturers use more stable forms, such as ascorbyl phosphate and vitamin E acetate.

Vitamin C is extremely difficult to maintain in premixes or feeds since it is susceptible to destruction by so many environmental factors, especially oxidation. Phosphorilation of ascorbic acid (Ascorbyl phosphate) produces a highly stable product.

Vitamin A Stability

New technology has further improved Vitamin A stability by a cross linking process, such as the reaction between the gelatin and the sugar, that makes the beadlet insoluble in water, giving it a more resistant coating that can sustain higher pressure, friction, temperature and humidity (see Fig. 18.7).

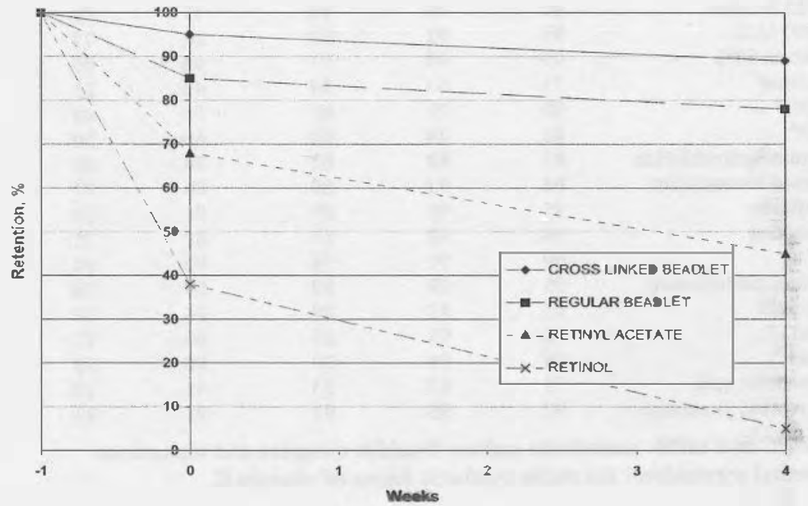


Figure 18.7 Stability of vitamin A on extrusion (37°C, 65% RH).

Stability studies conducted with new cross linked vitamin A indicate higher stability than with soluble beadlet. Chen (1990) measured the stability of the 3 cross linked vitamin A beadlets on the market in trace mineral premixes and feeds. After 3 months storage at high temperature and humidity, the vitamin A retention varied from 30 to 80%, depending on the antioxidant present in the beadlet. In a 30% dairy concentrate pelleted at 200°F, retention at pelleting varied from 78 to 96%. After three months storage at high temperature and humidity, retention varied between 57 and 62%. The improvements in vitamin A stability through extrusion, in the last decade, increased by 35% mainly due to the use of cross linking processing.

Table 18.16A Equivalent Average Retention of Vitamins During Pelleting as a Percent Retained (%) (BASF, 1992)

	Pelleting Temperature, °F/Conditioning Time, Min.				
Various equivalent combinations of temperature and time	140/2	160/2	180/2	200/2	220/2
	150/1	170/1	190/1	210/1	230/1
	160/0.5	180/0.5	200/0.5	220/0.5	
Vitamin	170/0.3	190/0.3	210/0.3	230/0.3	
A 650 beadlet	95	93	90	85	79
D_3 325 beadlet	97	95	93	91	89
D_3 400 M.S.	95	92	88	82	77
E acetate 50%	99	98	97	96	95
E alcohol	75	65	54	43	23
MSBC*	80	72	65	56	44
MPB*	82	74	68	60	50
Thiamin hydrochloride	93	89	82	74	63
Thiamin mononitrate	95	93	89	84	77
Riboflavin	95	93	89	84	78
Pyridoxine	94	92	87	82	75
B_{12}	99	97	96	95	94
Calcium pantothenate	95	93	89	84	78
Folic acid	95	93	89	84	77
Biotin	95	93	89	84	77
Niacin	96	94	90	86	80
C, Ascorbic acid	75	65	55	45	35
C, Ascorbyl phosphate	97	95	93	91	89

*MSBC and MPB: menadione sodium bisulfite complex and menadione dimethyl pyrimidinol are stable synthetic forms of vitamin K.

Choline

*MSBC and MPB: menadione sodium bisulfate complex and menadione dimethyl pyrimidinol are stable synthetic forms of vitamin K.

Table 18.16B Average Stability of Vitamins in Feed Stored at Room Temperature (BASF Corp., 1992)

	Vitamin retention (%)				
	Month				Loss/Month
Vitamin	0.5	1	3	6	%
A (beadlet)	92	83	69	43	9.5
D_3 (beadlet)	93	88	78	55	7.5
E acetate	98	96	92	88	2.0
E alcohol	78	59	20	0	40.0
MSBC	85	75	52	32	17.0
MPB	86	76	54	37	15.0
Thiamin hydrochloride	93	86	65	47	11.0
Thiamin mononitrate	98	97	83	65	5.0
Riboflavin	97	93	88	82	3.0
Pyridoxine	95	91	84	76	4.0
B_{12}	98	97	95	92	1.4
Calcium pantothenate	98	94	90	86	2.4
Folic acid	98	97	83	65	5.0
Biotin	95	90	82	74	4.4
Niacin	93	88	80	72	4.6
C, Ascorbic acid	80	64	31	7	30.0
C, Ascorbyl phosphate	95	90	83	75	4.5
Choline	99	99	98	97	1.0

Vitamin E and Minerals

Vitamin E, as dl-alpha-tocopherol, is an antioxidant by itself and, therefore, if applied directly to feeds, is consumed rapidly. The free phenolic hydroxy group in this molecule is responsible for the antioxidant activity. When the hydroxy group is protected by formation of an ester, as in tocopheryl acetate, the compound obtained is resistant to oxygen, since it has no double bonds or free hydroxy groups. Vitamin E acetate is stable in feeds with neutral or slightly acidic pH. However, even slightly alkaline conditions may affect the stability, such as when limestone carrier is used or in the presence of large quantities of magnesium oxide. Under these conditions, some of the protective acetate groups **split off and free tocopherol** is formed, which can be rapidly oxidized. Dove and Ewan (1986) determined the stability of alpha-tocopherol in feeds without and with trace minerals. At the end of 3 months storage at 25–30°C, alpha-tocopherol retention was 50% and 30%, respectively. The further addition of 245 ppm copper as copper sulfate, produced 0% retention after 15 days. Tocopherol, the most

concentrated form of vitamin E activity, is such an unstable vitamin form that it should not be considered for any animal nutrition application.

Schneider (1988) determined the stability of tocopheryl acetate and tocopherol in vitamin-trace mineral premixes stored at ambient and stressful conditions. At the end of I month storage at ambient conditions, the retention was 95% and 44%, respectively, and at high temperature and humidity, the retention was 90% and 13%, respectively.

VITAMIN K

Menadione, pure vitamin K_3, is a crystalline yellow powder that is unstable and irritating to skin and mucous membranes. It is not utilized in pure form, but is formulated with sodium bisulfate and derivatives thereof. Menadione sodium bisulfate complex (MSBC) and menadione dimethyl pyrimidinol bisulfate (MPB) are more stable than menadione.

B-VITAMINS STABILITY

B-vitamins are also unstable to a certain extent. Vitamin B_1 and B_6 are more stable under acidic conditions, while pantothenic and folic acids are most stable in a slightly alkaline environment. pH of the medium is far less important than the aggressiveness of moisture and trace elements. Thiamin hydrochloride is destroyed rapidly in a choline/trace mineral premix (high moisture, pH 4–5) while it is fairly stable in a basemix (low moisture, pH 7–8). Vitamin solubility in water is inversely correlated to stability. Thiamin mononitrate with a solubility of 10g/100mL, is significantly more stable in premixes than thiamin hydrochloride with a solubility of 100g/100mL (Adams, 1982), Fig. 18.8.

Vitamin B_6 is more rapidly destroyed in a choline chloride/trace mineral premix (high moisture) than in a basemix (low moisture). Calcium-d-panthothenate is quite stable. Losses occur only after prolonged storage at acidic pH. Riboflavin in stable in all premixes and also under climatic stress.

Vitamin B_{12} and choline are very stable compounds, but B_{12} is slightly sensitive to strong acid, alkali, reduction, light, ascorbic acid and ferrous sulfate.

Folic acid is stable to heat and air, but unstable in acid and alkaline solutions. It is light sensitive, slightly sensitive to moisture and sensitive to oxidation and reducing agents.

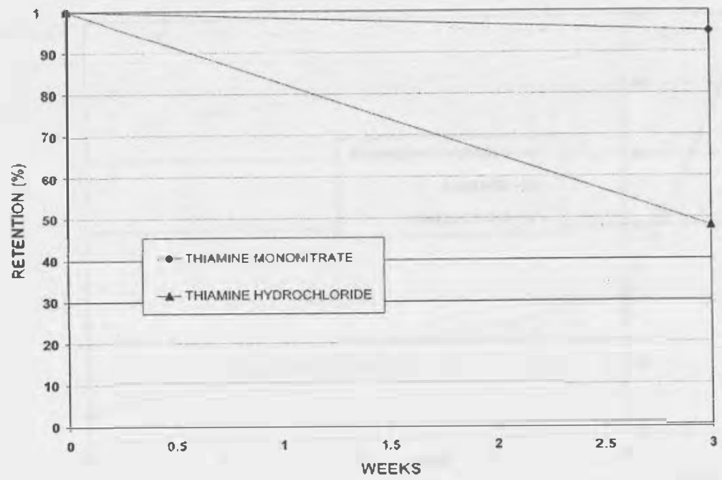


Figure 18.8 Storage stability of thiamine in a vitamin-T.M. Premix (110°F or 43°C, 85% RH; BASF Corp, 1992).

Zhuge and Klopfenstein (1985) determined the stability of riboflavin and niacin in a broiler premix without and with trace minerals. At the end of 7 months storage, riboflavin retained 50% and 46%, respectively. Niacin retained 96% and 91%, respectively. Schaaf (1990) reported retentions of 100%, 100% and 93% for pyridoxine, riboflavin and folic acid, respectively, in vitamin premixes stored at ambient temperature for 3 months. Christian (1983) in a basemix study, determined riboflavin and calcium pantothenate stability after 3 months storage. Riboflavin retained 72% at low temperature and humidity, and 35% at high temperature and humidity. Calcium pantothenate retained 52% and 16%, respectively. Adams (1982) reported the stability of pyridoxine and thiamin in premixes without and with trace minerals. After storage for 3 months under stressful conditions, pyridoxine retained 100% and 45%, respectively. After 21 days under stressful conditions, thiamin hydrochloride retained 48% and thiamin monoitrate, 95%. BASF (1986) compared the stability of crystalline ascorbic acid and ethyl cellulose coated ascorbic acid through pelleting. Crystalline retained 85% and ethyl cellulose, 82%. A follow up study determined the stability of ascorbyl-phosphate. This compound not only is very stable, but also maintains the bioavailability. Ascorbyl phosphate retained 95% through extrusion, Fig. 18.9.

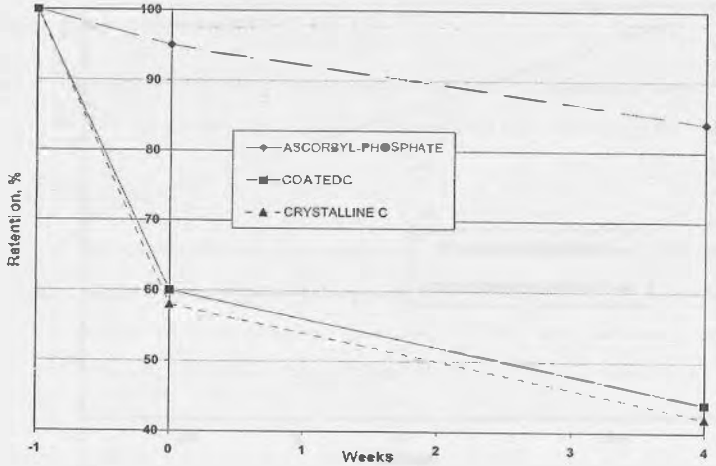


Figure 18.9 Stability of ascorbic acid on extrusion at a major catfish feed manufacturer (37°C, 65% RH; Source BASF Corp., 1992).

Extrusion manufacture increases digestibility of fish feeds over pelleted feeds. Additionally volatile components are flashed off the exudate. At the same time, vitamin stability is weakened during ingredient mixing, grinding, and exposure to high temperatures resulting from the extrusion process. The relative stability varies with the vitamin (see Table 18.17; Coelho, 1991).

Table 18.17 Stability of Vitamins to Feed Extrusion and Expected Vitamin Loss Per Month for the Various Classes of Vitamins (Coelho, 1991)

Very High	High	Moderate	Low	Very Low
Choline	Riboflavin	Thiamin	Thiamin	Menadione
Chloride		Mononitrate	HCl	
	Niacin	Folic Acid		Ascorbic Acid
Ascorbic				
Polyphosphate	Pantothenic	Pyridoxine		
Sulfate	Acid			Vit. E alcohol
Monophosphate	Vit. E acetate	Vit. D_3		
	Biotin	Vit. A		
	B_{12}			
1%/month	*6%/month*	*11%/month*	*17%/month*	*50%/month*

VITAMIN REQUIREMENTS AND STABILITY

To compensate for instability, feed manufacturers generally use relatively high levels of vitamin C supplementation (250–500 mg/kg), even though the minimum requirement is quite low (50–100 mg/kg). However, new protected or stabilized forms of vitamin C have been developed and are now commercially available. Recent advances in the stabilization of vitamin C made by Hoffman LaRoche, Pfizer and BASF, have resulted in chemically stabilized forms, ascorbic 2-polyphosphate, ascorbic 2-sulfate and ascorbic 2-monophosphate. These chemically stabilized forms of vitamin C are highly stable during extrusion manufacture. Earlier efforts to stabilize vitamin C using lipid encapsulation did not result in a product able to withstand the temperatures and moisture levels of extrusion manufacture. Commercial manufacture of aqua feeds should not rely on unstabilized forms of vitamin C (De Antonis et al. 1993).

While new forms of vitamin C have greatly improved the cost effectiveness of vitamin C fortification, over formulation of other vitamins for extruded aquafeeds is still commonly practiced today. Manufacturers generally add vitamins in excess of actual requirements to allow for losses during manufacture, shipping, and storage prior to feeding, referred to as overages. Overages for coldwater and marine species are higher than those for warmwater fishes such as tilapia. Further losses may occur in the water by leaching. For practical vitamin recommendations for supplemental overage allowances for tilapia and

warmwater fishes (see Table 18.18). These overages can be viewed as being a recommended supplemental vitamin addition to the natural vitamins that would be obtained by the fish from ingesting the rest of their diet.

18.13 FEEDSTUFF SELECTION

Feedstuffs for water restricted systems are selected based on unit costs for dietary energy, protein, and amino acid composition and ingredient digestibility and also by the level of phosphorus. Many potential feed ingredients can be eliminated from consideration based on their total phosphorus content. Of special concern are those ingredients that are high in ash and indigestible fiber. High ash ingredients such as some fishmeals, meat and bone meal, poultry meal, and soybean meal contain high amounts of calcium and phosphorus. Fish and animal bone and phytin are not completely digested and when discharged in feces contribute to phosphorus pollution of the water column. High levels of calcium and phytin-sequestered phosphorus have been implicated as a cause of cataracts in young salmonids and reducing the bioavailability of dietary zinc. Phosphorus in the form of phosphate is a contributor to eutrophication, and therefore should be controlled to the extent possible. The amount of phosphate entering the effluent can be reduced by purchasing feeds containing low levels of phosphorus.

Table 18.18 Recommended Supplemental Overage as a Percent for Specific Vitamins That are Used in Extruded Type Feeds Fed to Warmwater Fishes

Vitamin	Percent Overage
A - Acetate	150
D_3 - Cholecalciferol	130
E - Acetate	150
Thiamin mononitrate	250
B_{12}	130
Biotin	150
Folic Acid	200
Riboflavin	150
Niacin & Choline	150
Stabilized Vitamin C	110

Unpublished data, Paul Maugle (author)

The phosphorus/nitrogen (P/N) ratio of many ingredients such as animal by-product meals has been established. Many animal by-products meals have high P/N ratio. Generally, plant protein ingredients such as soybean and corn gluten meals have a lower P/N ratios, which is a desirable characteristic for inclusion in low environmental impact diet formulations. However, this beneficial characteristic is offset by other undesirable characteristics: indigestible phytin in the case of soybean meal and high levels of the carotenoid lutein associated with corn gluten meal preclude high inclusion levels in trout feeds.

An example of a derivative criteria, the P/N ratio of several ingredients provided by Cho et al. (1994) can be added to the nutrient specification matrix. The nutrient matrix will be discussed at a later point but it is important to know that actual nutrient levels as well as derivatives such as the P/N ratio can be used to assist in formulating low impact aqua feeds (see Table 18.9).

Ketola and Richmond (1994) have shown that rainbow trout requirements for dietary non-phytin phosphate are greater for bone mineralization than for weight gain. A summary of their findings, Table 18.19, suggest that the minimum level of non-phytin phosphorus in rainbow trout diets ranges between 0.51 and 0.61% depending on size of fish. As a safety margin, in commercial diets, actual levels are somewhat higher.

Table 18.19 Dietary Requirement for Non-phytin Phosphate in Trout (Ketola and Richmond, 1994)

	Small Trout	Large Trout
Maximum Growth	0.41%	0.34–0.54%
Maximum Bone Ash	0.51%	>0.54

Most discharge standards in the USA, with a few exceptions such as Idaho and New York, do not currently require low phosphorus discharge levels. However, future discharge standards may require a reduction in the amount of phosphates that are discharged. Additionally, aquaculture produced fish can and will continue to command increased values in the open market, due in part to the clean and wholesome image that has been created by recent marketing efforts, some of which emphasizes the benefits of RAS in particular to the environment.

ENERGY

The requirement for energy is an important aspect of fish nutrition. Fig. 18.10 shows the overall process of utilization of energy by fish. The

first fraction of energy loss occurs during the process of digestion, followed by losses through urinary and gill excretions and sloughing off of body tissues. During the process of digestion and absorption, there is an expenditure of energy called heat increment. The energy that is retained is called net energy. Part of the net energy is used for metabolism and voluntary activity, leaving that which is actually recoverable in tissue growth and reproduction.

Overall, the major nutrients in fish feed (protein, fat, and carbohydrates) vary in their digestible energy contents, Table 18.20. The digestible energy content of protein is about 4.5 kilocalories per gram, when digestibility is about 90%. The digestible energy content of fat is about 8 to 9 kcal/g. Values for carbohydrates are variable. For instance, starch, which is a complex carbohydrate, has a fairly high gross energy

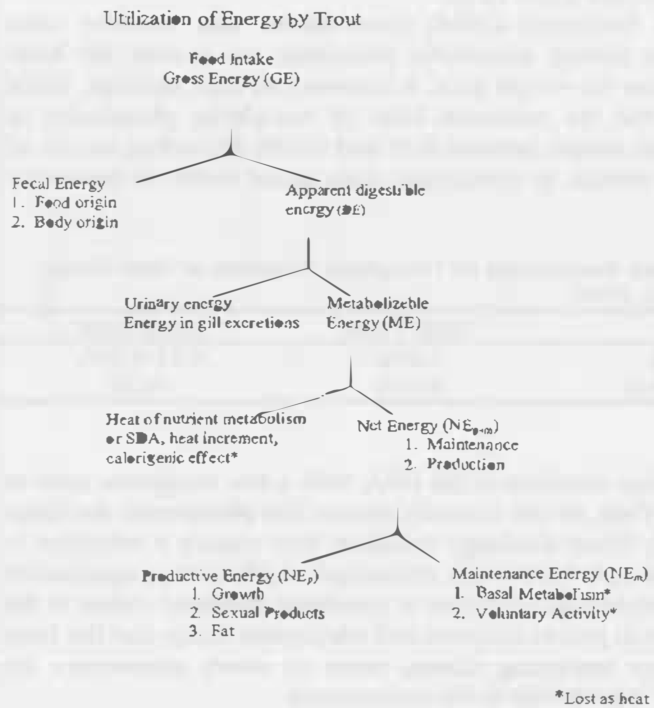


Figure 18.10 Overall process of utilization of energy by fish.

content (4.18 kcal/g), but its digestibility decreases as the level of starch increases in the diet, thus its digestible energy also decreases. Simple sugars do not require digestion, are readily absorbed and therefore they have high digestible energy values regardless of level in the feed.

Fish Oils

For diets that include oil as a dietary component, fish oil is still the major oil component, although other oil sources can be substituted depending upon the fish species, e.g., tallow, white grease, poultry fat, and soy oil. However, the successful use of fish oil requires special standards and criteria to be implemented. In the commercial arena, a key to quality is to purchase cold processed fish oil. Upon arrival of fish oil at the mill, additional antioxidants should be added. The addition of 250–500 ppm butylated hydroxyanisole (BHA), butylated hydroxytoluene (BHT), or other antioxidant is necessary to assure the stability of fish oil through the manufacturing process and subsequent shipment to the end user. These antioxidants can be added by the fish oil supplier and / or by the feed manufacturer at ingredient reception but long term stability of oils in the feed requires its utilization. Fish oils that are acceptable for use in fish feeds should contain less than 3% free fatty acids, less than 1% moisture, less than 1% nitrogen and less than 20% Totox defined as (2 X (peroxide value) + (Anisidine Value)). Fish oil should not be stored in heated tanks as elevated storage temperature can contribute to increased oxidation.

Table 18.20 Major Nutrients in Fish Feed (Protein, Fat, and Carbohydrates) vary in their Digestible Energy Contents

Nutrient Class		Digestibility	Energy (kcal/g) Gross	Digestible
Protein		90[1]	–	4.5[2]
Fat: (PUFA)		90[3]	9.3	8.4
Carbohydrate[3]				
Starch	20%	69	4.18	2.9
	40%	53	4.18	2.2
	60%	26	4.18	1.1
Glucose	20%	99	3.72	3.7
	40%	99	3.72	3.7
	60%	99	3.72	3.7
Sucrose	20%	99	3.94	3.9
	40%	99	3.94	3.9
	60%	99	3.94	3.9

[1] Cho & Slinger (1979) [2] Available energy, Smith (1971) [3] Singh & Nose (1967)

ESSENTIAL FATTY ACIDS

Fish require specific fatty acids in their diets. Fish, as a rule, require n-3 fatty acids, i.e., eicosapentaenoic (20:5 n3), docosahexaenoic (22:6 n3), or linolenic acid (18:3 n3) (NRC, 1983). Some tilapia may require only n-6 fatty acids such as linoleic (18:2 n6) and arachidonic acids (20:4 n6) (Takeuchi et al. 1983; and Kanazawa et al. 1980). Common carp and chum salmon (*Oncorhynchus keta*) may require n-3 and n-6 fatty acids (Watanabe et al. 1975; and Takeuchi and Watanabe, 1982). Generally, fatty acids should compose about 1% of the diet. Deficiency signs include reduced growth and sometimes fatty liver degeneration, anemia, fin erosion, increased susceptibility to bacterial infection such as flexobacteria, and fainting syndrome or stress shock when agitated.

PROTEIN REQUIREMENTS

Figure 18.11 shows the results of feeding coho salmon (*Oncorhynchus kisutch*) a series of diets containing no carbohydrate and varying ratios of protein and fat (Ketola, H.G., unpublished). These data demonstrate a protein/energy relationship in the diet. As the protein increases up to 54%, the growth rate of coho salmon significantly increased. Beyond that level, however, there was no further benefit from increasing the level of protein. Also, fat and protein contents of the

carcass shows marked changes as the ratios of dietary protein/fat changed. The optimum fat and protein content in the carcass is probably that associated with the minimum protein needed for maximum growth (54%). Above that level, the deposition of fat is further decreased, and the fish becomes leaner.

The protein requirements of various species of fishes are shown in Table 18.21. The so-called "protein requirement" is a summation of amino acid requirements that must be provided to the fish. There are about 10–12 amino acids that are essential to different species of fish. Other non-essential amino acids serve as non-specific nitrogen sources in the synthesis of other amino acids and proteins. Each species has a rather specific ratio of essential amino acids. Deficiency of any one of these amino acids causes a reduction in the rate of growth. Also, excesses of some amino acids caused reduced growth. Therefore, the balance of amino acids is important. There are two amino acids that are unique: methionine and phenylalanine. While these amino acids are essential, there are two other amino acids, which can substitute for part of their needs. Cysteine and cystine can replace part of the need for methionine, and tryosine can replace part of the need for phenylalanine. The information on the requirements for amino acids is not extensive.

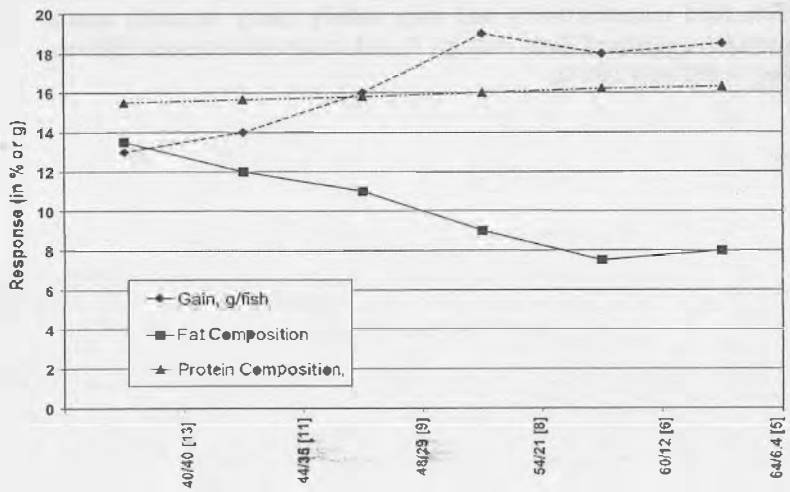


Figure 18.11 Effect on weight gain and composition of Coho Salmon (*Oncorhynchus kisutch*) fed diets containing varying ratios of protein and fat (Ketola, H.G., unpublished).

Table 18.22 summarizes quantitative amino acid requirements for several species of fish. A few specific signs of deficiencies have been observed. For instance, deficiency of lysine causes caudal fin erosion (Ketola, 1979). Deficiency of methionine causes cataracts (Poston et al. 1967). Deficiency of tryptophan causes cataracts and scoliosis (Poston and Rumsey, 1983). For further information on protein and amino acid requirements and deficiencies, see reports by Cowey and Sargent (1972), Halver (1989), Lovell (1989), Stickney and Lovell (1977), MAFES (1982) and NRC/NAS (1981, 1983).

When a researcher simply requires a diet for adequate laboratory research, probably the best thing to do is purchase an appropriate premixed feed from a commercial feed manufacturer. There are many such diets available for catfish, trout, and salmon. For names of commercial fish feed manufacturers, consult advertisements in Salmonid (U.S. Trout Farmers Association, Harpers Ferry, WV), Northern Aquaculture (Victoria, B.C., Canada) or Aquaculture News (Jonesville, LA) or other aquaculture magazines or newspapers. When feed of a specifically defined composition is required, there are several federal specification diets such as the ASD and Abernathy federal formulations (see Tables 18.23 and 24). These diets may be also be purchased from several fish feed manufacturers and may satisfy many research needs. Compositions for purified diets may be found in several sources (Halver, 1989; NRC, 1981 and 1983).

Table 18.21 Protein Requirements of Various Species of Fish

Species (Scientific name)	Requirement (% of diet)
Rainbow trout (*Oncorhynchus mykiss*)	40-46
Chinook salmon (*O. tshawytscha*)	40
Plaice (*Plauronectas platessa*)	50
Guilthead bream (*Sparus aurata*)	50
Common carp (*Cyprinus carpio*)	31–38
Channel catfish (*Ictalurus punctatus*)	31–38
Japanese eel (*Anguilla japonica*)	32–36
Grass carp (*Ctenopharyngodon idella*)	44.5
Puffer fish (*Fugu rubripes*)	50
Malabar grouper (*Epinephelus malabaricus*)	40–50
Milkfish (fry) (*Chanos chanos*)	40
Red sea bream (*Pagrus major*)	55
Smallmouth bass (*Micropterus dolomieu*)	45
Largemouth bass (*Micropterus salmonides*)	40
Tilapia	
Tilapia aurea (fry)	56
Tilapia aurea	34
Tilapia mossambica	40
Tilapia zillii	35

NRC/NAS (1981; 1983)

There are some species and life stages (early, swimup) that cannot be successfully maintained by feeding a typical trout, salmon, or catfish diet. For these cases, you may choose among many excellent aquarium fish foods that are available in a variety of forms such as flakes, pellet, crumbles, and natural and live food organisms. Some species may require a special formula feed that can be developed with the aid of a nutritionist, feed manufacturer or aquarium fish specialists. Others may require a closer imitation of their natural foods.

Table 18.22 Quantitative Amino Acid Requirements for Several Species of Fishes

Amino acid	Salmon	Eel	Carp	Catfish	Trout
	Species				
Arginine	6.0	4.5	4.2	4.3	5.9
Histidine	1.8	2.1	2.1	1.5	1.6
Isoleucine	2.3	4.0	2.3	2.6	2.1
Leucine	4.0	5.3	3.4	3.5	3.7
Lysine	5.0	5.3	5.7	5.0	6.1
Methionine & Cysteine	3.8	5.0	3.1	2.3	3.0
Phenylalanine & Tyrosine	5.3	5.8	6.5	5.0	3.1
Threonine	2.3	4.0	3.9	2.0	3.4
Tryptophan	0.5	1.1	0.8	0.5	0.6
Valine	3.2	4.0	3.6	3.0	2.2

Recommendations based upon: NRC (1981; 1983). Trout: Ketola (1983), Ogino (1980), Poston and Rumsey (1983), Rumsey et al. (1983), Hughes et al. (1983).

Table 18.23 Federal Specified Diet for Atlantic salmon ASD2-30 (for Granules and Pellet Sizes)

1.	Fish food shall carry the following guaranteed analysis:	
	Crude protein,	≥ 55.0%
	Fish meal protein	≥ 33.0%
	Crude fat	≥ 17.0%
	Moisture (at sack-off)	≤ 10.0%
2.	Herring meal	≥ 50%
3.	Dried shrimp meal: minimum protein 38%	5.0%
4.	Soy flour: defatted, minimum protein 48.5%, maximum fat 1%	20.3%
5.	Dried blood flour, minimum protein 80%	10%
6.	Trace mineral premix No. 2 (USFWS)	0.5%
7.	Vitamin premix No. 30 (USFWS)	0.6%
8.	Choline chloride, 50%	0.22%
9.	Ascorbic acid	0.075%
10.	Herring oil: stabilized with 0.04 BHA-BHT (1:1) or 0.01% ethoxyquin	12.0%
11.	Lignin sulphonate pellet binder, e.g., Ameribond, Orzan, or equivalent	2.0%
12.	Fish oil stabilized with 0.04% BHA-BHT (1:1) or 0.01% ethoxyquin OR soybean lecithin may be used at no more than 2% with fish oil	12.0%

Table 18.24 Federal Specifications: Abernatby Diet S8-2 (84) (for Starter Crumbles)

1.	Fish food shall carry the following guarantee:	
	Crude protein, not less than 48%	≥48.0
	Fish meal protein, not less than 40.5%	≥ 40.5
	Crude fat, not less than 17% or more than 19%	≥ 17.0
	Moisture, not more than 10% at sack-off	≤ 10.0
2.	Herring meal, minimum protein 70%, 8–12% fat, maximum salt 3%, maximum ash 15%. Stabilized	50%
3.	Dried whey product or dried whey, minimum protein 12%	10%
4.	Spray dried blood flour or flash dried blood meal	10%
5.	Condensed fish solubles, minimum protein 30% OR poultry byproduct meal, protein 60% to 68%, fat maximum 12%, ash maximum 16%	1.5%
6.	Wheat standard middlings, wheat mill run, or wheat shorts	Remainder
7.	Vitamin premix No. 2 (USFWS)	1.5%
8.	Choline chloride, 60% product	0.58%
9.	Ascorbic acid	0.1%
10.	Trace mineral mixture No. 1 (USFWS)	0.5%
11.	Lignin sulphonate pellet binder	2%
12.	Fish oil stabilized with 0.04% BHA-BHT (1:1) or 0.01% ethoxyquin OR soybean lecithin may be used at no more than 2% with fish oil	12%

18.14 Feedstuff Digestibility

Feed should be selected with the thought in mind that the feed will contain indigestible nutrients that become either biological waste products, or which contribute to nutrient pollution because they are not completely digested. Ammonia, nitrate, phosphate, and organic matter are some of the primary dissolved pollutants. With that in mind, feeds should meet the nutritional requirements of the species for optimal growth while at the same time minimizing nutrient excess. Feeds formulated with excessive protein or made with poorly digestible ingredients should be avoided as much as possible. Though fish can utilize protein for energy, this approach results in increased ammonia production. Net protein utilization (NPU) is the term used to describe the amount of body protein gain divided by protein fed. NPU can be improved by reducing the amount of protein used for energy metabolism. This is accomplished by optimizing the protein to energy ratio of the feed. Improvement of NPU can also be accomplished through feeding an

ideal protein or a protein source of high biological activity, preferably both. Feeding an ideal protein presents an amino acid pattern that best meets the species requirements.

Differences in digestible energy of key ingredients used in commercial fish feeds are summarized in Table 18.25 for trout and Table 18.26 for catfish and tilapia (note each fish species is different). The ingredients selected for use in trout feeds significantly impact the digestibility (Kaushik and Medale, 1994) of the finished feed.

The digestibility of carbohydrates varies among species and the degree of digestibility of different feed components can be quite wide (see Table 18.27 for trout). Raw or partially cooked carbohydrates are more easily digested by catfish and tilapia than by salmonids. However, undigested carbohydrates increase the quantity of feces excreted. During the feed manufacturing process, the cooking of starches during extrusion improves the durability and water stability of the pellet, and can reduce the amount of solid waste produced.

A number of management practices can also affect the digestibility of a feed. As meal and feed pellet size increases, digestive and absorptive efficiencies decrease (Solomon and Brafield, 1972 and Windell et al. 1978). More frequent feedings of smaller meals tend to increase the digestibility of a given diet formulation.

Table 18.25 Apparent Protein Digestibilityof Ingredients in Diets for Trout

Ingredient	Digestible Protein (%DM)	Digestible Energy (kJ/g DM)
Fish Meal	66.1	19.4
Corn Gluten Meal	58.9	17.5
Soy Bean Meal	46.5	13.5
Triticale	14.4	13.6
Rapeseed	39.6	14.9

*4.186 kJ = 1 kcal

Table 18.26 Apparent Protein Digestibility of Ingredients in Diets for Catfish and Tilapia

Ingredient Digestible Energy (Mcal/g)		
Ingredient	Catfish	Tilapia
Alfalfa, 17% Protein	0.67	1.01
Corn Grain		
Raw	1.10	2.46
Processed	2.53	3.02
Menhaden Fish Meal	3.90	4.04
Molasses	3.47	2.94
Soybean Meal, 48% Protein	2.58	3.34
Wheat Flour	2.55	2.89

Table 18.27 Apparent Digestibility of Carbohydrate in Ingredients in Diets for Trout

Ingredient	ADC of Starch (%)
Rice	39
Wheat	54
Potato	<5
Tapioca	<15
Corn	33
Extruded Wheat Starch	96

Many of the differences in ingredient or nutrient digestibility between species are a function of whether a particular species possesses an endogenous source of certain digestive enzymes (Maugle, 1982). At present, there are a number of commercially available exogenous enzymes that can increase the availability of nutrients of select ingredients. Trout are not insulin deficient and are able to utilize mono and diglycerides. In preliminary studies with extruded trout feed made by Southern States, a 0.2 (as 1.2 reduced to 1.0) Feed Conversion Unit improvement was found in fish reared with hemicellulase and xylase supplements, (ChemGen) when compared to trout reared with the same diet but without the enzyme supplement.

METHODS OF EVALUATION DIGESTIBILITY

There is no more accurate way to determine how a particular feed is suited for your management style and particular strain of fish than to perform some simple digestibility studies. Depending upon your staff qualifications, you can do this internally or arrange to have a trained fish nutritionist help you with the study (such assistance can often can be arranged through your feed supplier). The basic methodology of this study is that a non-digestible marker, such as chromic oxide (Cr_2O_3) is included in the diet at a low concentration (known value, e.g., 1% by weight). The amount of the marker going through the GI tract will remain fixed as mass, but other nutrients will be extracted by the fish as the feed is digested, thus increasing the percentage concentration of the marker in the feed. Working with your feed manufacturer, you can then use this data to customize a feed to your operation. See the following detailed example.

APPARENT DIGESTIBILITY IN FISH

Apparent digestibility of a nutrient is the net digestibility of the nutrient. Thus, these two terms, apparent and net, are used interchangeable. We will define apparent digestibility $D_{\%}$, mathematically as:

$$D_{\%} = 100 \cdot \frac{Amount\ of\ nutrient\ fed - Amount\ of\ nutrient\ in\ feces}{Amount\ of\ nu\ rient\ fed} \tag{18.2}$$

$D_{\%}$ can be determined by use of indicators without measuring feed intake or excreta output. Indicators may be natural indigestible constituents in feed or they may be added to it. Suitable indicators should:

- be totally indigestible and unabsorbable
- be easy to assay
- not be synthesized by the animal
- pass through the digestive tract at the same rate as feed
- be biologically inert so that its presence in the feed should not influence digestion or physiology.

A number of digestibility indicators have been proposed such as lignin or acid-insoluble ash. Added indicators include chromic (III) oxide (Cr_2O_3), ferric oxide or oxides of rare earths. Chromic oxide is commonly used.

If the concentrations of the indicator substance [Cr] and a nutrient are determined for a feed and for representative samples of unleached excreta, then the $D_{\%}$ of the nutrient can be calculated from the following equation:

$$D_{\%} = 100 - 100 \cdot \left(\frac{Cr\ in\ feed}{Cr\ in\ excreta}\right) \cdot \left(\frac{Nutrient\ in\ excreta}{Nutrient\ in\ feed}\right) \tag{18.3}$$

One way to obtain unleached excreta from aquatic animals such as fish is by manual stripping, Austreng (1978). For example, fish should be fed the test diets containing the indicator (chromic oxide) for sufficient time to ensure normal consumption and complete replacement of all other non-indicator feed residues in the digestive tract. Fish are netted one at a time, and firmly but gently held in a moistened towel or glove while gentle pressure is applied to the abdominal region of starting just behind the pelvic fins and progressing toward the anus to expel excreta. Uncontaminated (without milt or urine) excreta are collected. If excreta are not readily expelled, the fish are rejected. The procedure should not be done on abnormal trout that show obvious signs of reduced growth, general weakness or lethargy. Samples of excreta should be collected from enough fish and pooled until the amount is sufficient for required analyses. Samples of feed and excreta are analyzed for concentration of indicator [Cr] and [nutrient] of interest. For further information about determination of apparent digestibility, the reader is referred to Austreng (1978), Bondy (1987), and Halver (1989).

EXAMPLE

As an example of calculating $D_{\%}$, suppose a diet containing 5.4% nitrogen (N) and 0.5% indicator chromium (as Cr_2O_3) was fed to trout for 10 days after which excreta were sampled, dried, and analyzed for the percentage concentrations of N and indicator. Results of the analyses and calculations for apparent digestibility of nitrogen and dry matter are shown in Table 16.28 below:

Table 18.28 Example Data for Collecting the Percentage Apparent Digestibility of Nitrogen and Dry Matter

Trout	Cr [indicator] (%)		N [nitrogen] (%)	
Feed	Feed	Excreta	Feed	Excreta
Test feed	0.5	1.8	5.4	2.0

Calculations:

$$D_{\%DM} = 100 - 100 \cdot \left(\frac{Cr\ in\ feed}{Cr\ in\ excreta}\right) = 100 - 100 \cdot \left(\frac{0.5}{1.8}\right) = 72.2\%$$

$$D_{\%N} = 100 - 100 \cdot \left(\frac{Cr\ in\ feed}{Cr\ in\ excreta}\right) \cdot \left(\frac{Nutrient\ in\ excreta}{Nutrient\ in\ feed}\right)$$

$$= 100 - 100 \cdot \left(\frac{0.5}{1.8}\right) \cdot \left(\frac{2.0}{5.4}\right) = 89.7\%$$

In this example, the apparent digestibility of dry matter ($D_{\%DM}$) is 72.2% and that for dietary nitrogen ($D_{\%N}$) is 89.75.

Note, an example calculation demonstrating how to determine if there is a significant difference in mean results is given in the Appendix.

18.15 Pelleted, Expanded, and Extruded Feeds

Fish feeds today are manufactured in three types: extruded, expanded, or steam pellets. Extruded and expanded pellets have improved durability and digestibility, in comparison to steamed. Extruded pellets can be produced to be heavier or lighter than water, providing excellent management options to the culturist. Extruded feed typically produces feed with the best feed conversion numbers as well as providing the most uniform product in size/shape/quality. Expanded pellets are about as good as extruded pellets and at times are very difficult to tell apart from extruded. Steam pelleting is preferable when adding medication, but is generally not as desirable because of its limitations concerning buoyancy and feed conversion. There is considerable variability between the feeds of different manufacturers in important feed characteristics, such as grind size, digestibility, and the amount of fines. The grind size should be no larger than 1.2 mm (3/64-inch) for general fish diets and no larger than 0.8 mm (2/64-inch) for larval diets (one way to compare quality and price among manufacturers is the grind size). A key mistake that many make is to think the cost of the pellet is the most important cost parameter. This is not so; rather, the important cost parameter is the cost to produce a kg of fish. The most expensive feed to buy may produce fish growth at the lowest total cost when conversion and water quality, which affects feed conversion, are

considered. The culturist should create a strong working relationship with feed manufacturer, sales representative, and nutritionist.

> **"Rule of Thumb"**
> The lowest priced feed usually does not produce the lowest feed cost per unit gain.

Each pelleting process can be defined as to its advantages and disadvantages. Today, most aquaculture feeds are purchased as extruded pellets, primarily it seems for the ability to create a floating pellet. This is not necessarily the best choice; it depends upon your farm. Review your options closely before deciding. One of the main disadvantages of the expanded feed pellet is that they slowly sink. RAS operators often prefer the floating pellet because the high turbidity in RAS's usually means you cannot "see" what is happening very far down into the water column. Cornell successfully used a slow sink expanded pellet for many years in clear but dark water in a trout RAS.

You should probably pay more attention to the overall quality of the feed and particularly the percent of fines than whether it is a sinking or floating pellet. Perhaps using a combination of floating and sinking would be a good alternative. Remember, fish adapt slowly going from one type of feed to another in terms of sinking or floating.

Steam Pelleting

Steam pelleting is the oldest of the three methods. After the ground mash is premixed, steam is added to condition and partly gelatinize the starches and to assist in pellet binding. The mash is typically exposed to steam for short periods of time (less than 35 seconds), and to processing temperatures of 100–180°F (38–82°C). The conditioned mash is forced through constricting tapered holes in a die and the resulting pellets are cut by a series of knives to a desired length. After the pellets are cut, they are blown dry with a final moisture content of 9–10%. Steam pellets are cylindrical in shape typically having lengths 1.5 to 2 times longer than their diameter. They are hard and have a glassy exterior. Steam pellets are fairly dense and will sink; their advantages are:

- Less energy is required during manufacture.
- Less heat means less destruction of heat sensitive nutrients, medications, and vitamins.
- Initial cost is less.

Now the disadvantages of steam pelleting:

- Greater incidence of fines.
- Pellets sink.
- Smallest sized pellet available is roughly 2.4 mm (only good for larger fish).
- Total fat content cannot exceed 20%.
- Marginal pellet durability in the water column.

EXTRUDED

Extruded feeds are produced using a pre-mixed mash introduced into an extruder barrel and a significant amount of water being added. This high moisture mash is then placed under intense pressure, heat and friction. This causes starch gelatinization two to three times that found in steam pelleting. Some manufacturers precondition the mash to improve palatability, digestibility, and durability. Processing temperatures during extrusion can reach up to 300°F (149°C). This super heated mixture is then forced through a die. This causes a rapid reduction in pressure, which in turn causes the pellets to expand. The expansion reduces pellet density that creates a pellet that is less dense than water, i.e., a floating pellet. Pellets leaving the die have moisture contents 10–15% higher than steam pellets, which then requires additional energy to dry the product to 10% moisture. Advantages of extruded feed are:

- Expansion can be controlled to allow the product to sink, slow sink, or float (allows fish management options to observe feeding behavior).
- Fat levels higher than 20% are possible (allows higher energy feed pellets).
- Higher temperature process improves availability of nutrients (arguable) resulting in better feed conversion ratios.
- Better digestibility means less waste for the system to handle.
- Carbohydrates are used as binders so "nutritionally blank" ingredients are not required to bind the product. Better feed conversions and reduction in total fecal load is the result.
- Structural integrity allows for smaller, more consistent sizes.
- Pellets are durable, uniform in size, and have few fines.

Everything is not a plus using extruded feeds, disadvantages are:

- Nutrient, medication, and vitamin degradation is higher because of the additional heat used in the manufacturing process. This requires that these ingredients be supplemented at higher levels.
- Costs are higher to the purchaser due to higher manufacturing equipment costs, the need for nutrient supplementation, and a slower production time (average cost of 0.2 to 5% higher than steam pelleted feed).
- Truck hauling costs are higher since pellet density is lower (typical bulk trucks might be able to hold 40,000 pounds (18,180 kg) of extruded feed compared to 42,000 pounds (19,090 kg) of steam pelleted feed).

Rangen Feeds (Buhl, Idaho) and Melick Feeds (Catawissa, PA) are two companies that manufacture an extruded feed.

EXPANDED

The expanded feed manufacturing process is similar to that of extruded feed except the cooked mash must be sent through a pellet mill to form the pellets, which typically results in sinking pellets (rarely and with great skill, it is possible to produce a floating feed). Expansion does not require as much moisture as extrusion, which permits the pellets to be dried without heat, which reduces operating costs. Expanders produce denser pellets than extruders but not as dense as a steam pellet. There are only a few expanders currently in use in the United States to produce fish feed (Zeigler Brothers, Gardners, PA is an example).

18.16 FLOATING FEEDS

Whether a feed pellet floats, partially sinks, or sinks is determined by the level of starch in the formula as well as the degree of striation of the starches and the bulk density of the exudate. For example, a 20.5% starch diet will yield a slowly sinking feed in which approximately 50% of feed initially floats when first immersed in water but sinks within minutes of being fed. In general, maintaining the diet between 20 to 22% starch will promote a floating pellet and support the proper formation of a feces

casing[3] for fish that produce a casing around their feces, e.g., tilapia. Diets that are too high in protein will result in diarrhea.

The floating or sinking characteristics as well as pellet durability can be adjusted by the selection of feedstuffs, the amount of water and oil used internally on the mix, and the amount of cook during extrusion. The amount of water and oil added internally to the feed can either enhance or inhibit the striation of starches. In general, a higher starch diet will reduce feces specific weight; thus making the feces more apt to float. This knowledge can be applied proactively to work in your favor. For example, if dissolved air flotation is a primary means of removing feces and suspended solids, then a higher starch diet would be an advantage. On the other hand, in an RAS that uses a central drain system for feces and sedimentation removal, a higher protein and lower starch diet would be advantageous, as the feces would tend to sink.

Table 18.29 Comparisons of Feed Manufacturing Techniques

	Compressed Pellets	Extruded	Expanded
Initial feed cost	lowest	highest	intermediate
Starch gelatinization, %	<40	>80	60–80
Max. temp., °F (°C)	180 (82)	300 (149)	300 (149)
Max. fat level, %	20	40	30
Digestibility	good	best	better
Sinking available	yes	yes	yes
Floating available	no	yes	possibly
Slow sink available	no	yes	possibly
Fines upon receipt, %*	1 to 6	<1	<1
Vitamin and nutrient degradation	lowest	highest	intermediate
Feed conversions	worst	best	intermediate
Uniformity of feed	variable	excellent	good
Availability	most mills	some mills	few mills

*Fines can result from poor handling practicing of finished feed or minimal grinding

[3] The casing around a fecal strand is a glycoprotein that is a mixture of starch and protein. That is why the starch protein ratios have to be maintained within some range in order to produce a casing that will retain its form.

18.17 Summary

Summaries of the three feed manufacturing processes and their key characteristics are given in Table 18.29. Use it as a guide and not a bible. Fish performance as affected by the feed will depend upon a lot of things besides the type of pellet.

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CHAPTER 19

AQUAPONICS: INTEGRATING FISH AND PLANT CULTURE[1]

19.0 INTRODUCTION

Aquaponics, the combined culture of fish and plants in recirculating systems, has become increasingly popular. Now there is even a news group (send an email to: snsaquasys@townsqr.com - type subscribe in subject line) on the Internet that discusses many aspects of aquaponics on a daily basis. Since 1997, a quarterly periodical (*Aquaponics Journal*) has published informative articles, conference announcements and product advertisements. At least two large suppliers of aquaculture and/or hydroponic equipment have introduced aquaponic systems to their catalogs. Hundreds of school districts are including aquaponics as a learning tool in their science curricula. At least two short courses on aquaponics have been introduced, and the number of commercial aquaponic operations, though small, is increasing.

Aquaponic systems are recirculating aquaculture systems that incorporate the production of plants without soil. Recirculating systems are designed to raise large quantities of fish in relatively small volumes of water by treating the water to remove toxic waste products and then reusing it. In the process of reusing the water many times, non-toxic nutrients and organic matter accumulate. These metabolic byproducts need not be wasted if they are channeled into secondary crops that have economic value or in some way benefit the primary fish production system. Systems that grow

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additional crops by utilizing by-products from the production of the primary species are referred to as integrated systems. If the secondary crops are aquatic or terrestrial plants grown in conjunction with fish, this integrated system is referred to as an aquaponic system.

Plants grow rapidly in response to dissolved nutrients that are excreted directly by fish or generated from the microbial breakdown of fish wastes. In closed recirculating systems with very little daily water exchange (less than 5%), dissolved nutrients accumulate and approach concentrations that are found in hydroponic nutrient solutions. Dissolved nitrogen, in particular, can occur at very high levels in recirculating systems. Fish excrete waste nitrogen directly into the water through their gills in the form of ammonia. Bacteria convert ammonia to nitrite and then to nitrate. Ammonia and nitrite are toxic to fish, but nitrate is relatively harmless and is the preferred form of nitrogen for growth of higher plants, such as fruiting vegetables. It is the symbiotic relationship between fish and plants that makes the consideration of an aquaponic system a reasonable system design criteria.

Aquaponic systems offer several advantages. In RAS's, the disposal of accumulated waste is always a major concern. Recirculating systems are promoted as a means of reducing the volume of waste discharge to the environment. Certainly the volume is reduced but the pollution load (organic matter, dissolved nutrients) per unit of discharge is correspondingly higher. This more concentrated discharge may pose a threat to the environment in some situations, or an additional expense if the wastewater is discharged to a municipal sewer system for further treatment. Effluent is discharged from the system to eliminate organic sediment and prevent nutrient buildup.

In aquaponic systems, the plants recover a substantial percentage of these nutrients, thereby reducing the need to discharge water to the environment and therefore extending water use, i.e., by removing dissolved nutrients through plant uptake, the water exchange rate can be reduced. Minimizing water exchange reduces operating costs of aquaponic systems in arid climates and heated greenhouses where water or heated water represents a significant expense. Lennard (2006) demonstrated that nitrate accumulation in culture waters was reduced by up to 97% (Table 19.1) in the Aquaponic system when compared with the fish-only system.

Profitability is always a major concern when considering a recirculating system. Recirculating systems are expensive to construct and operate, and profitability often depends on serving niche markets for live fish such as tilapia, whole fresh fish on ice, or other high value products. A secondary plant crop, which receives most of its required

nutrients at no additional cost, improves system profit potential. The daily feeding of the fish provides a steady supply of nutrients to plants, which reduces or eliminates the need to discharge and replace depleted nutrient solutions or adjust nutrient solutions as is required in hydroponics. The carbon dioxide vented from fish culture water can increase plant yields in enclosed environments. The plants purify the culture water and can, in a properly sized and designed facility, eliminate the need for separate and expensive biofilters. Biofiltration represents a major capital expense and a minor operational expense. In well-designed aquaponic systems, the hydroponic component can provide sufficient biofiltration for the fish, and therefore the cost of purchasing and operating a separate biofilter is avoided. These costs are charged to the hydroponic subsystem, which, in the case of lettuce, generates approximately two thirds of the system's income. The profitability of recirculating systems can thus be improved substantially with aquaponics, if there is a good market for the vegetable crop.

Table 19.1 Fish Growth, Lettuce Yield and Nitrate Removal for Fish-only Systems and Aquaponic Systems (Lennard, 2006)

Parameter	Fish-only	Aquaponic
Fish FCR	0.87 ± 0.01	0.88 ± 0.0
Lettuce yield (kg/m^2)	NA	5.77 ± 0.19
NO_3 accumulation (mg/l)	52.20 ± 5.28	1.43 ± 1.09
NO_3 removal (%)	0	97

The expense of water quality monitoring is reduced in aquaponic systems as waste nutrients are generated daily at uniform levels and there is generally excess wastewater treatment capacity. An aquaponics system also generates savings in several areas of construction and operation by sharing operational and infrastructural costs for pumps, blowers, reservoirs, heaters, and alarm systems. Initial capital investment is reduced in that an aquaponics system can be erected with a modest increase in acreage over that required for a hydroponic facility. Aquaponic systems do require high capital investment, moderate energy inputs, and skilled management. The premium prices available in niche markets may be required for an aquaponic facility to be profitable.

There are, of course, disadvantages to aquaponic systems. The most obvious of these is the large ratio of plant growing area in comparison to

the fish rearing surface area. A large ratio of plant surface to fish surface is needed to achieve a balanced system where nutrient levels stay relatively constant. For example in the UVI raft system the ratio of plant growing area to fish surface area ratio is 7.3. Larger ratios arc needed as solids removal efficiency decreases. In essence, aquaponic systems emphasize plant culture, which is an advantage if viewed by a horticulturist. Most of the labor expended in the facility is devoted to seeding, transplanting, maintaining, harvesting, and packing plants. Additionally, a new set of skills is required for the plant component, so a commercial operation would do better with both an aquaculturist and horticulturist on staff. Another disadvantage is that the horticulturist must rely on biological control methods rather than pesticides to protect the plants from pests and diseases. However, this restriction can be viewed as an advantage in that the plant products can be niche marketed as "pesticide free".

19.1 System Design

The design of aquaponic systems closely mirrors that of recirculating systems in general with the addition of a hydroponic component and the possible elimination of a separate biofilter and devices (foam fractionators) for fine and dissolved solids removal. Fine solids and dissolved organic matter generally do not reach levels that require foam fractionation in aquaponic systems at the recommended design ratio. The essential elements of an aquaponic system consist of a fish rearing tank, a settleable and suspended solids removal component, a biofilter, a hydroponic component and a sump, Figure 19.1 (Rakocy and Hargreaves, 1993).

Effluent from the fish rearing tank is treated first to reduce organic matter concentration in the form of settleable and suspended solids. Next, the culture water is treated to remove ammonia and nitrite by fixed-film nitrification, which often occurs in the hydroponic component. As water flows through the hydroponic unit, some dissolved nutrients are recovered by plant uptake. Finally, water collects in a reservoir (sump) where it is returned to the rearing tank. The location of the sump may vary. For example, if elevated hydroponic troughs are used, the sump can be located after the biofilter and water would be pumped up to the troughs and returned by gravity to the fish rearing tank.

The system can be configured such that a portion of the flow is diverted to a particular treatment unit (Naegel, 1977; Wren, 1984). For example, a small side-stream flow may go to a hydroponic component

after solids removal, while most of the water passes through a biofilter and returns to the rearing tank.

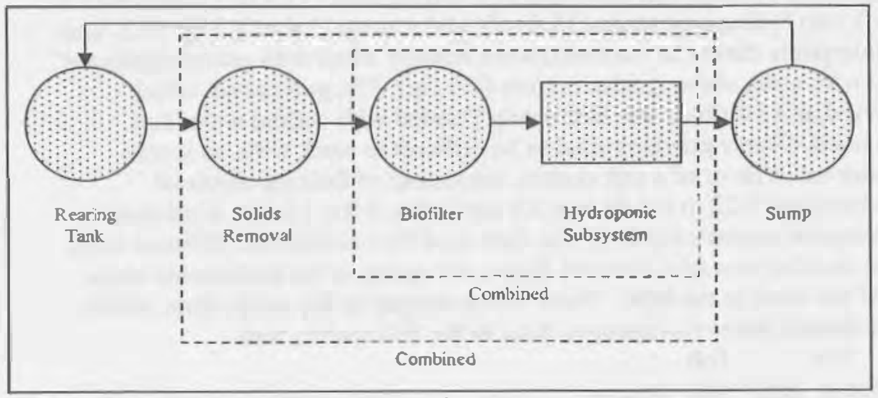


Figure 19.1 Optimum arrangement of aquaponic system components.

The biofiltration and hydroponic components can be combined by using a plant support media, such as gravel, (Lewis et al. 1978; Sutton and Lewis, 1982; Rakocy, 1984; Watten and Busch, 1984) or sand (McMurtry et al. 1990), which also functions as biofilter media. Raft hydroponics, which consists of floating sheets of polystyrene and net pots for plant support, can also provide sufficient biofiltration if the plant production area is sufficiently large (Rakocy, 1995). Combining biofiltration with hydroponics is a desirable goal because eliminating the expense of a separate biofilter is one of the main advantages of aquaponics. An alternative design combines solids removal, biofiltration, and hydroponics in one unit. The hydroponic support media (pea gravel) captures solids and provides surface area for fixed-film nitrification, although with this design it is important not to overload the unit with suspended solids. An overload of suspended solids is always a threat due to variations in fish feeding activities and efficiency of the solid removal component. For these reasons, gravel or sand beds should be avoided for large commercial-scale operations.

Aquaponics Research at the University of the Virgin Islands (UVI)

Aquaponics research at the University of the Virgin Islands (UVI) has focused on the culture of tilapia in outdoor tanks equipped

with raft hydroponics. As the UVI system developed, there were many design evolutions. Most of the experimental work was conducted in six replicated systems that consisted of a rearing tank (12.8 m^3), clarifier (1.9 m^3), two hydroponic tanks (13.8 m^2), and a sump (1.4 m^3), Fig. 19.2. The hydroponic tanks (28 cm deep) were initially filled with gravel supported by wire mesh above a false bottom (7.6 cm). The gravel bed, which served as a biofilter, was alternately flooded with culture water and drained. Coarse gravel proved to be difficult to work with, so it was removed in favor of a raft system, consisting of floating sheets of polystyrene 1.22 m x 2.44 m x 3.8 cm (4 ft x 8 ft x 1.5 in). A rotating biological contactor (RBC) was then used for nitrification. Effluent from the clarifier was split into two flows, one going to the hydroponic tanks and the other to the RBC. These flows merged in the sump, from which the treated water was pumped back to the fish rearing tank.

The fish rearing tank was situated under an opaque canopy and the clarifier and sump were covered with plywood. Shading inhibits algae growth, lowers daytime water temperature, and creates more natural light conditions for the fish. The rearing tank in this particular design proved to be too large relative to the plant growing surface area of the hydroponic tanks, or, conversely, the hydroponic tanks were too small relative to the size of the rearing tank. When the rearing tank was stocked with tilapia at commercial densities (107 fish/m^3), the daily feed ration to the system was so high that nutrients rapidly accumulated to levels that exceeded the recommended upper limits for hydroponic nutrient solutions (2,000 mg/L as total dissolved solids, TDS) (Rakocy et al. 1993). The optimum ratio between the fish feeding rate and plant growing area was determined, using Bibb lettuce as the baseline plant, to be 57 g of feed/day/m^2 of plant growing area, (Rakocy, 1989a). At this ratio, the nutrient accumulation rate decreased, and the hydroponic tanks were able to provide sufficient nitrification. This success enabled the RBCs to be removed when the fish stocking rates were reduced to levels that allowed feed to be administered near the optimum rate for good plant growth. The optimum ratio will vary depending on plant species and the production method, i.e., staggered vs. batch culture.

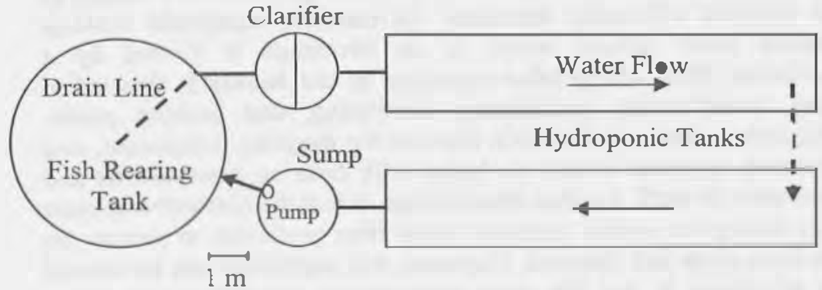
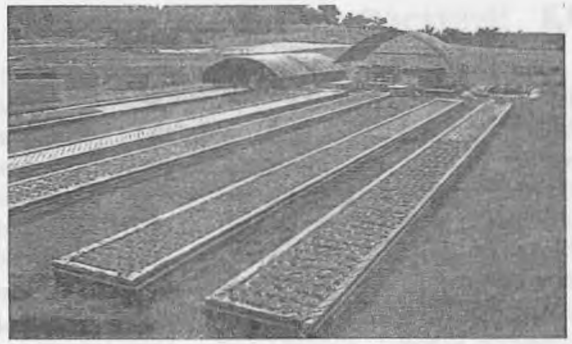


Figure 19.2 Design of UVI experimental aquaponic system.



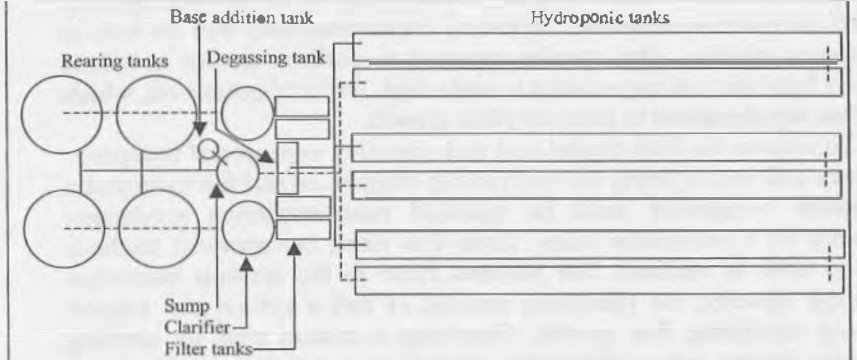
The experimental system was scaled up two times. In the first scale-up, the length of each hydroponic tank was increased from 6.1 m to 29.6 m. The optimum design ratio of 57g feed/day/m^2 of plant growing area allowed the rearing tank to be stocked with tilapia at commercial levels (for a diffused aeration system) without excessive nutrient accumulation.

"Rule of Thumb"

57 g of feed/day per square meter of plant growing Area for staggered production of Bibb lettuce

In the second scale-up, the number of hydroponic tanks (29.6 m in length) was increased to six and the number of fish rearing tanks was increased to four, Fig. 19.3. This production unit design represents a realistic commercial scale, although there are many possible size options

and tank configurations. For example, the number of hydroponic tanks could be reduced from six to two, as in the experimental unit, by increasing the width or length as long as the total area remained the same. Likewise, the rearing tanks (7.8 m^3) could be increased in size and/or number, provided there is a corresponding increase in the surface area of the hydroponic tanks. There is more flexibility in sizing and configuring outdoor systems, which are restricted to tropical or subtropical climates for year-round production of fish, than is available for indoor systems. In temperate indoor systems, the components must be configured tightly to conserve space, and the unit cannot exceed the width of a greenhouse bay, which ranges from 6.7 to 9.45 m. UVI's commercial-scale unit (second scale-up) could be configured to occupy as little as 0.05 ha of land.



TANK DIMENSIONS

Rearing tanks: Diameter: 10 ft, Height: 4 ft, Water volume: 2,060 gal each?
Clarifiers: Diameter: 6 ft, Height of cylinder: 4 ft, Depth of cone: 3.6 ft, Slope: 45°, Water volume: 1,000 gal
Filter and degassing tanks: Length: 6 ft, Width: 2.5 ft, Depth: 2 ft, Water volume: 185 gal
Hydroponic tanks: Length: 100 ft, Width: 4 ft, Depth: 16 in, Water volume: 3,000 gal, Growing area: 2,304 ft^2
Sump: Diameter: 4 ft, Height: 3 ft, Water volume: 160 gal
Base addition tank: Diameter: 2 ft, Height: 3 ft, Water volume: 50 gal
Total system water volume: 29,375 gal
Flow rate: 100 GPM,
Water pump: ½ hp
Blowers: 1½ hp (fish) and 1 hp (plants)
Total land area: 1/8 acre

PIPE SIZES

Pump to rearing tanks: 3 in
Rearing tanks to clarifier: 4 in
Clarifiers to filter tanks: 4 in
Between filter tanks: 6 in
Filter tank to degassing tank: 4 in
Degassing to hydroponic tanks: 6 in
Between hydroponic tanks: 6 in
Hydroponic tanks to sump: 6 in
Sump to pump: 3 in
Pipe to base addition tank: ¾ in
Base addition tank to sump: 1.25 in

Figure 19.3 Layout of UVI aquaponic system with tank dimensions and pipe sizes.

19.2 FISH PRODUCTION

Tilapia is the most common fish cultured in aquaponic systems. Although some aquaponic systems have used channel catfish, largemouth bass, crappies, rainbow trout, pacu, common carp, koi carp, goldfish, Asian sea bass (barramundi) and Murray cod, most commercial

systems are used to raise tilapia. The majority of freshwater species, which can tolerate crowding, including ornamental fish, will do well in aquaponic systems. One species reported to perform poorly is hybrid striped bass because they cannot tolerate high levels of potassium, which is often supplemented to promote plant growth.

To recover the high capital cost and operating expenses of aquaponic systems and earn a profit, the fish rearing component and the hydroponic vegetable component must be operated near maximum production capacity on a continuous basis. Three fish stock management methods can be used to maintain fish biomass close to the systems maximum carrying capacity, the maximum amount of fish a system can support without restricting fish growth. Operating a system near its carrying capacity utilizes space efficiently, maximizes production, and reduces variation in the daily feed input to the system, an important factor in sizing the hydroponic component. The basic methods of fish management are sequential rearing, stock splitting and multiple rearing units. It is important to determine the best method for varying circumstances prior to designing a commercial system, as each method has different requirements. Changing rearing methodology in a production mode is costly and interruptive to steady fish production

Sequential Rearing

Sequential rearing involves the culture of several age groups (multiple cohorts) of fish in the same rearing tank. When one age group reaches marketable size, it is selectively harvested with nets and a grading system, and an equal number of fingerlings are immediately restocked in the same tank. There are three problems with this system: 1) the periodic harvests stress the remaining fish and could trigger disease outbreaks; 2) stunted fish avoid capture and accumulate in the system, wasting space and feed; 3) it is difficult to maintain accurate stock records over time, which leads to a high degree of management uncertainty and unpredictable harvests. Nevertheless, sequential rearing has been used successfully on a commercial scale with tilapia, a very hardy species, despite its drawbacks. Sequential rearing may be risky for species less tolerant to the repeated stress of partial harvest.

Stock Splitting

Stock splitting involves stocking very high densities of fingerlings and periodically splitting the population in half as the carrying capacity of the rearing tank is reached (Van Gorder, 1991). A typical stock splitting system would divide the initial population three times so that the fish would go from one tank to two tanks and then from two tanks to four tanks and finally from four tanks to eight tanks, from which marketable fish are harvested. Alternatively, single cohorts can be moved to successively larger a tank, which reduces stress on the fish and generally takes less time than trying to physically divide a single cohort into two equal populations, based on weight or number, to be placed in tanks of the original size.

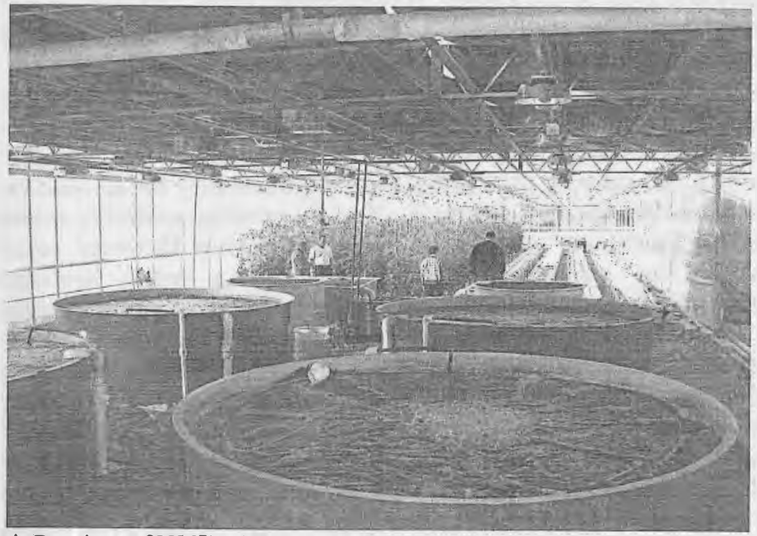
If the splitting cohort technique is chosen, a total of 15 rearing tanks are required to be able to harvest eight tanks at the end of each growth interval, a period of 5 or 6 weeks for tilapia. This method of fish management avoids the carryover problem of stunted fish and improves the accuracy of stock inventory assessment. However, the moves can be very stressful on the fish unless a swimway is installed, which connects all the rearing tanks to a narrow channel and the fish can be herded into it through a hatch in the wall of the rearing tanks and maneuvered into another rearing tank by movable screens. With swimways, the division of the populations in half involves some guesswork because the fish cannot be weighed or counted. An alternative method involves crowding the fish with screens and pumping them into a hauling tank or directly into another tank using a pescalator.

Multiple Rearing Units

With multiple rearing units, the entire population is moved to larger rearing tanks when the carrying capacity of the initial rearing tank is reached. The fish are either herded through a hatch between adjoining tanks or into "swimways" connecting distant tanks. Multiple rearing units usually come in modules of two to four tanks and are connected to a common filtration system. After the largest tank is harvested, all of the remaining groups of fish are moved to the next largest tank, and the smallest tank is restocked with fingerlings.

A variation of the multiple rearing unit concept is the division of a long raceway into compartments with movable screens. As the fish grow, their compartment is increased in size and moved closer to one end of the raceway where they will eventually be harvested. These should be cross-flow raceways or mixed-cell raceways (see Chapter 4) to ensure uniform water quality throughout the length of the tank. In a cross-flow raceway influent water enters the raceway through a series of ports down one side of the raceway while effluent water leaves the raceway through a series of drains down the other side. This system ensures that water is uniformly good throughout the length of the raceway.

Another variation is the use of several tanks of the same size. Each rearing tank contains a different age group of fish, but they are not moved during the production cycle. This system does not utilize space efficiently in the early stages of growth, but the fish are never disturbed and the labor involved in moving the fish is eliminated.



A Portion of UVI's Aquaponic System Showing (counterclockwise from upper right): Clarifier, Two Filter Tanks, Degassing Tank with Internal Standpipe Well, and Return Sump.

UVI's current commercial-scale, aquaponic system uses multiple rearing tanks to simplify stock management, as the fish are not moved during their 24-week growout cycle. The system consists of four fish rearing tanks (7.8 m^3 each, water volume), two cylindro-conical clarifiers (3.8 m^3 each) with a 45° slope, four filter tanks (0.7 m^3 each), one degassing tank (0.7 m^3), six hydroponic tanks (11.3 m^3 each, 29.6 m x 1.2 m x 0.4 m), one sump (0.6 m^3) and one base addition tank (0.2 m^3). The total hydroponic surface area is 214 m^2, and the total system water volume is 110 m^3. Tilapia production is staggered in the four rearing tanks so that one rearing tank is harvested every 6 weeks. At harvest the rearing tank is drained and all of the fish are removed. The rearing tank is then refilled with the same water and immediately restocked with fingerlings for a 24-week production cycle

The system is used to culture Nile tilapia (*Oreochromis niloticus*) and red tilapia. Fry are sex-reversed with 17α-methyltestosterone according to the INAD (Investigations in New Animal Drugs) protocol to obtain a consistently high percentage (~99%) of male fingerlings. Nile tilapia fingerlings are stocked at a rate of 77 fish/m^3 to obtain a harvest size of 800–900 g. These large fish are processed for the fillet market. Red tilapia fingerlings are stocked at 154 fish/m^3 to achieve an average weight near 500 g for the whole fish West Indian market. Every six weeks approximately 500 to 600 kg of fish are harvested. Annual production has been 9,152 lbs. (4.16 mt) for Nile tilapia and 10,516 lbs (4.78 mt) for red tilapia (Table 19.2). However, production can be increased to 11,000 lbs (5 mt) with close observation of the *ad libitum* feeding response.

Tilapia grow well at high densities if good water quality is maintained. In the commercial-scale system, dissolved oxygen (DO) levels in the rearing tanks are maintained at 5–6 mg/L by high DO in the incoming water and by diffused aeration with air delivered through 22 air stones around the perimeter of the tank. A 1.5-hp (1.1 kW) blower provides air to the rearing and degassing tanks. Vigorous aeration vents carbon dioxide gas into the atmosphere and prevents its buildup. A high water exchange rate quickly removes suspended solids and toxic waste metabolites (ammonia and nitrite) from the rearing tank. A ½-hp in-line pump produces a flow of 380 Lpm (100 gpm) and an average retention time of 1.4 hours/rearing tank. However, flows to the individual rearing tanks are adjusted so that the tank with the highest biomass receives the highest flow rate, which exceeds 130 Lpm (35 gpm) for a retention time of less than 1 hour. The other rearing tanks receive proportionately lower flow rates relative to their biomass. Values of ammonia-nitrogen and nitrite-nitrogen in the rearing tanks are approximately 1–2 mg/L and <1 mg/L, respectively.

Through careful attention to management of the water quality parameters of DO, ammonia-nitrogen and nitrite-nitrogen, it has been possible to grow tilapia at high densities. Other water quality variables of importance to the system are water temperature, pH, and alkalinity. Water temperature ranges from a low of 23.0°C in the winter to a high of 29.0°C in the summer. The average water temperature has been 27.0°C, which is lower than the optimum temperature (30°C) for tilapia and higher than the optimum temperature (20–22°C) for many vegetables. The system water temperature is lower than that in nearby ponds because none of the system's surface area is exposed to direct sunlight. The pH is generally maintained at 7.0 by adding equal amounts calcium hydroxide

and potassium hydroxide. Total alkalinity averages approximately 100 mg/L as calcium carbonate ($CaCO_3$).

Table 19.2 Average Production Values for Male Mono-sex Nile and Red Tilapia in the UVI Aquaponic System. Nile Tilapia Are Stocked at (77 fish/m^3 (0.29 fish/gallon) and Red Tilapia Are Stocked at (154 fish/m^3 (0.58 fish/gallon).

Tilapia	Harvest Weight per Tank (lbs)	Harvest Weight per Unit Volume (lb/gal)	Initial Weight (g/fish)	Final Weight (g/fish)	Growth Rate (g/day)	Survival (%)	FCR
Nile	1,056 (480 kg)	0.51 (61.5 kg/m^3)	79.2	813.8	4.4	98.3	1.7
Red	1,212 (551 kg)	0.59 (70.7 kg/m^3)	58.8	512.5	2.7	89.9	1.8

In general, it is recommended that the carrying capacity in aquaponic systems should not exceed 60 kg/m^3 (0.50 lb/gallon). This density will promote fast growth and efficient feed conversion and reduce crowding stress that may lead to disease outbreaks. Pure oxygen is generally not needed to maintain this density.

The logistics of working with both fish and plants are challenging. In the UVI system one rearing tank is stocked every 6 weeks. Therefore 18 weeks are required before a system is fully stocked. If multiple units are used, then fish may be stocked as frequently as once weekly and harvested and sold once weekly. Similarly, staggered crop production requires frequent seeding, transplanting, harvesting, and marketing. Therefore, the overarching goal in the design process is to reduce labor requirements wherever possible and make operations as simple as possible. For example, the purchase of four fish rearing tanks adds extra expense. One larger tank could be purchased instead and partially harvested and partially restocked every six weeks. However, this operation requires additional labor, which is a recurring cost, and makes management more complex. In the long run having smaller, multiple tanks, in which the fish are not disturbed until harvest (hence, less mortality and better growth) will be more cost effective.

19.3 Solids

Fish generate fecal waste, most of which should be removed from the waste stream before it enters the hydroponic tanks. Other sources of particulate waste are uneaten feed and organisms (e.g., bacteria, fungi, and algae) that grow in the system. If this organic matter accumulates in the system, it will depress dissolved oxygen (DO) levels as it decays and produce carbon dioxide and ammonia. If deep deposits of sludge form, they will decompose anaerobically (without oxygen) and produce methane and hydrogen sulfide, which is very toxic to fish.

Suspended solids have special significance in aquaponic systems. Suspended solids entering the hydroponic component may accumulate on plant roots and produce a deleterious effect by creating anaerobic zones and blocking the flow of water and nutrients to the plant. However, some accumulation of solids may be beneficial. As solids undergo decomposition by microorganisms, inorganic nutrients essential to plant growth are released to the water, a process known as mineralization. Mineralization supplies several essential nutrients. Without sufficient solids for mineralization, more nutrient supplementation is required, thereby increasing the operating expense and management complexity of the system. However, it may be possible to minimize or eliminate nutrient supplementation if fish stocking and feeding rates are increased relative to plants. Another benefit of solids is brought about by the action of decomposing microorganisms. Microbes associated with decomposing solids are antagonistic to plant root pathogens and help maintain healthy root growth. Therefore, it appears that a delicate balance must be reached between excessive accumulation of suspended solids and insufficient accumulation.

Chapter 5 Solids Capture describes some of the common devices used for removing solids from recirculating systems. These include settling basins, tube or plate separators, the combination particle trap and sludge separator, centrifugal separators, microscreen filters and bead filters. Sedimentation devices (e.g., settling basins, tube or plate separators) primarily remove settleable solids (>100 microns) while filtration devices (e.g., microscreen filters, bead filters) remove settleable and suspended solids. Solids removal devices vary in regards to efficiency, solids retention time, effluent characteristics (both solid waste and treated water) and water consumption rate. While many devices may be appropriate for aquaponic systems, there is no research on the relationship between techniques for solids removal and the performance of hydroponic vegetables.

Sand and gravel hydroponic substrates are sometimes used to remove solid waste from the water flow stream. The solids remain in the system to provide nutrients to the vegetables through mineralization. As solids accumulate in the media, there is an increase in the cation exchange capacity (CEC), i.e., the ability of the media to adsorb and retain cations, positively charged nutrients, which are available for plant growth. Since cation concentrations are often high in aquaponic systems, CEC is generally not an important factor to plant growth. The use of sand is becoming less common, but one popular aquaponic system uses small beds 2.4 m by 1.2 m (8 ft by 4 ft) containing pea gravel ranging from 3 to 6 mm (1/8 to 1/4 inch) in diameter. The hydroponic beds are flooded several times daily with system water and then allowed to drain completely, and the water returned to the rearing tank. During the draining phase, air is brought into the gravel. The high oxygen content of air (compared to water) speeds the decomposition of organic matter in the gravel. The beds are inoculated with worms (*Eisenia foetida*), which improve bed aeration and assimilate organic matter.

The organic waste produced in aquaponic systems does not break down completely. The fraction of organic waste that resists microbial decomposition is referred to as being refractory. Particulate refractory compounds will slowly accumulate in substrates such as pea gravel or on the bottom or raft system troughs. Dissolved refractory compounds give the culture water a brown or tea color, which contains tannic acid, humic acid and other humic substances. These compounds have mild antibiotic characteristics and are beneficial to the system's fish and plants. Humic compounds form metalo-organic complexes with Fe, Zn, and Mn and thereby increase the availability of these micronutrients to plants.

SOLIDS REMOVAL

The most appropriate device for solids removal in a particular system may depend primarily on the organic loading rate (daily feed input and feces production) and secondarily on the plant growing area. For example, if large amounts of fish (high organic loading) are raised relative to the plant growing area, then a highly efficient solids removal device such as a microscreen drum filter is desirable. Microscreen drum filters capture fine organic particles, e.g., 60 micron and larger, which are retained by the screen for only a few minutes prior to backwashing and removal from the system. In this system, the dissolved nutrients excreted directly by the fish or produced by mineralization of very fine particles and dissolved organic matter may be sufficient for the size of the plant growing area. At the other extreme, if small amounts of fish (low organic

loading) are raised relative to the plant growing area, then solids removal may be unnecessary, as more mineralization is needed to produce sufficient nutrients for the relatively large plant growing area. However, un-stabilized solids, solids that have not undergone microbial decomposition, should not be allowed to accumulate on the tank bottom and form anaerobic zones. A reciprocating pea gravel filter (subject to flood and drain cycles), in which incoming water is spread evenly over the entire bed surface, may be the most appropriate device in this situation because solids are evenly distributed in the gravel and exposed to high oxygen levels (21% in air as compared to 0.0005-0.0007% in fish culture water) on the drain cycle, thereby enhancing microbial activity and increasing the mineralization rate.

UVI's commercial-scale aquaponic system relies on two cylindro-conical clarifiers to remove settleable solids (Rakocy, 1984). The fiberglass clarifiers each have a volume of 1.9 m^3 (500 gallons). The cylindrical portion of the clarifier is situated above ground and has a central baffle that is perpendicular to the incoming water flow (Figure 19.4). The lower conical portion has a 45° slope and is buried below ground. A drainpipe is connected to the apex of the cone. The drain pipe rises vertically out of the ground to the middle of the cylinder and is fitted with a ball valve. Rearing tank effluent enters the clarifier just below the water surface. The incoming water is deflected upward by a 45° pipe elbow to dissipate the current. As water flows under the baffle, turbulence diminishes and solids settle on the sides of the cone. The solids accumulate there and form a thick mat that eventually rises to the surface of the clarifier. To prevent this, approximately 30 male tilapia fingerlings are required to graze on the clarifier walls and consolidate solids at the base of the cone. Solids are removed from the clarifier three times daily. Hydrostatic pressure forces solids through the drain line when the ball valve is opened. A second, smaller baffle keeps floating solids from being discharged to the filter tanks.

The fingerlings serve another purpose. They swim into and through the drain lines and keep them clean. Without tilapia, the 10.2 cm (4 in) drain lines would require manually cleaning nearly every day due to bacterial growth in the drain lines, which constricts water flow. A cylindrical screen attached to the rearing tank drain prevents fingerings from entering the rearing tank.

In UVI's current commercial-scale system, the clarifier volumes have been increased to 3.8 m^3 (1000 gal) to achieve a 20-minute retention time, which resulted in much more effective removal of settleable solids. The clarifiers were constructed with standard fiberglass material. The slope of the conical portion was reduced to 45°. Although a

60° slope is more efficient for solids removal, installation of a heavy fiberglass tank with such a large cone is not feasible. Each clarifier is stocked with 40 male tilapia fingerlings to enhance clarifier performance, e.g., for grazing on surfaces and resuspending solids from upper reaches of the clarifier.

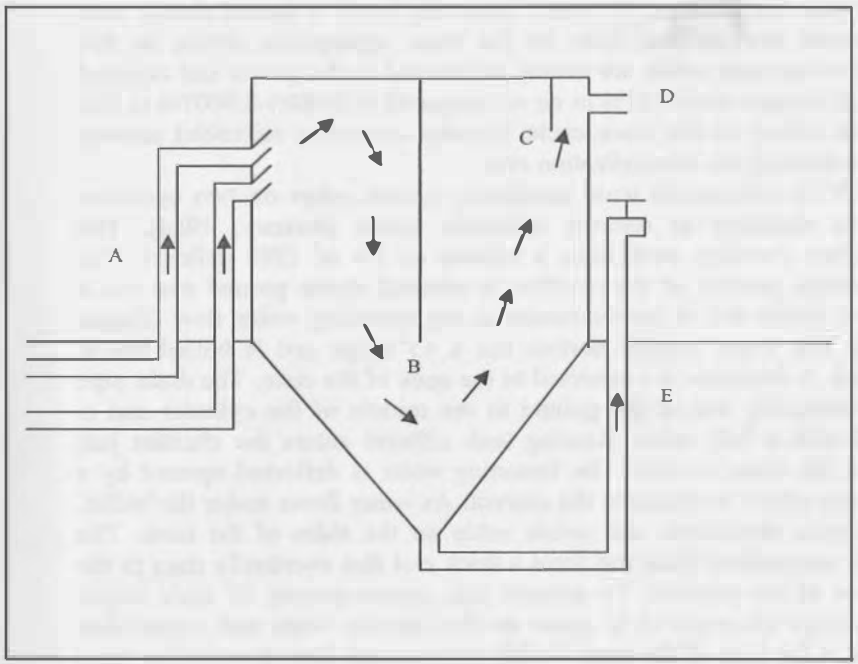


Figure 19.4 Cross sectional view (not to scale) of UVI clarifier showing drain lines from two fish rearing tanks (A), central baffle (B) and discharge baffle (C), outlet to filter tanks (D), sludge drain line (E) and direction of water flow (arrows).

Although the cylindro-conical clarifier can remove 21% of the dry weight of feed added to the system during a production cycle (Rakocy et al. 1991), large quantities of solids are not removed. Twarowska et al. (1996) determined that 35.3% of feed input to a tilapia culture system can be captured as settleable and non-settleable solids (based on volatile solids analysis) by using a particle trap and particle separator (17.6% removal) in combination with a microscreen drum filter (17.7% removal). These results indicate that the clarifier removes approximately 59% of the total removable solid waste. Although

fingerlings are needed for effective clarifier performance, their grazing and swimming activities are also counterproductive in that they resuspend some solids which exit through the clarifier outlet. The fish in the clarifier grow rapidly and should be replaced with small (~50 g) fingerlings every 12 weeks.

With clarification as the sole method of solids removal, large quantities of solids were discharged into the hydroponic tanks where they settled out and formed sludge deposits more than 5-cm deep at the influent end. This was a very undesirable condition, adversely affecting the plant component, as the sludge would float to the surface, engulf the plant roots and either kill the plants or greatly reduce their growth. A series of experiments resulted in arriving at a design solution that incorporated another water treatment stage consisting of additional tanks filled with orchard netting for the removal of fine solids. Two rectangular filter tanks 0.7 m^3 (185 gal) were installed after each clarifier. Effluent from the clarifier flows through these tanks in series. The netting is washed once or twice a week with a high-pressure water sprayer, and all the water in the filter tanks is discharged and the sludge is discharged to lined holding ponds. Prior to cleaning, a small sump pump is used to carefully return the filter tank water to the rearing tanks without dislodging the solids. This process conserves water and nutrients.

Effluent from the UVI rearing tanks is highly enriched with dissolved organic matter, which stimulates the growth of filamentous bacteria in the drain line, clarifier, and screen tank. The bacteria appear as translucent, gelatinous, light-tan filaments. Tilapia consume the bacteria and controls its growth in the drain line and clarifier, but bacteria does accumulate in the filter tanks. Without the filter tanks, the bacteria would overgrow plant roots. The bacteria do not appear to be pathogenic, but they do interfere with the uptake of dissolved oxygen, water, and nutrients, thereby affecting plant growth. The feeding rate to the system and the flow rate from the rearing tank determine the extent to which filamentous bacteria manifests itself, but it can be contained by

providing a sufficient area of orchard netting, either by adjusting screen tank size or using multiple screen tauks. In systems with lower organic loading rates (i.e., feeding rates) or lower water temperature (hence, less biological activity), filamentous bacteria diminish and are not a problem.

The organic matter that accumulates on the orchard netting between cleanings forms a thick sludge. Anaerobic conditions develop in the sludge, which leads to formation of gases such as hydrogen sulfide, methane, and nitrogen. Therefore, a degassing tank is used in the UVI system to receive the effluent from the filter tanks. A number of air diffusers vent the gasses into the atmosphere before the culture water reaches the hydroponic plants. The degassing tank has an internal standpipe well that splits the water flow into three sets of hydroponic tanks.

In UVI's commercial-scale system, the combination clarifier and filter tanks have maintained very low levels of suspended solids. In one trial with leaf lettuce, suspended solids values averaged 4.2 mg/L in the filter tank effluent (Rakocy et al. 1997). The hydroponic tanks also contribute to suspended solids removal. There is no phytoplankton in the water, which is clear but darkly colored with humic substances

Solids that are discharged from aquaponic systems must be disposed in an environmentally acceptable manner (see Chapter 6 Solids Management). There are several methods for effluent treatment and disposal. Effluent can be stored in aerated ponds and applied as relatively dilute sludge to land after organic matter has stabilized. This method is advantageous in dry areas where sludge can be used to irrigate and fertilize field crops. The solid fraction of the sludge can be separated from the water and used with other waste using geotextile membranes and polymers and used with other water products from the system (vegetable matter) to form compost. As an example, solids from the UVI commercial-scale aquaponic system are discharged through drain lines into two lined 16-m^3 ponds, which are continuously aerated with diffused air. As one pond is being filled over a 2 to 4-week period, water from the other pond is used to irrigate and fertilize field crops. Urban area facilities might have to discharge solid waste into sewer lines for disposal at the municipal sewage treatment plant.

19.4 BIOFILTRATION

A major concern in aquaponic systems is the removal of ammonia, a metabolic waste product excreted through the gills of fish. Ammonia will accumulate and reach toxic levels unless it is removed by the process of

nitrification (referred to more generally as biofiltration), in which ammonia is oxidized first to nitrite, which is toxic. Then nitrite is oxidized to nitrate, which is relatively non-toxic. Two groups of naturally-occurring bacteria (*Nitrosomonas* and *Nitrobacter*) mediate this two-step process. Nitrifying bacteria grow as a film (referred to as biofilm) on the surface of inert material or they adhere to organic particles. Biofilters contain media with large surface areas for the growth of nitrifying bacteria. Aquaponic systems have used biofilters with sand, gravel, shells, or various plastic media as substrate (Rakocy and Hargreaves, 1993). Biofilters perform optimally at a temperature range of 25–30°C, a pH range of 6.0–9.0, saturated DO, low BODs < 20 mg/L, and total alkalinity of 100 mg/L or greater. Nitrification is an acid-producing process that destroys alkalinity. Therefore, an alkaline base must be added frequently depending on feeding rate to maintain relatively stable pH values. Some means of removing dead biofilm is necessary to prevent media clogging, short circuiting of water flow, decreasing DO values and declining biofilter performance. A discussion of nitrification principles and a description of various biofilter designs and operating procedures are given in Chapter 7 and 8.

Major biofilter options (rotating biological contactors, expandable media filters, fluidized bed filters, and packed tower filters) are reviewed in Chapter 7 Biofiltration. If a separate biofilter is required or if a combined biofilter (biofiltration and hydroponic substrate) is used, the standard equations used to size biofilters may not apply to aquaponic systems as additional surface area is provided by plant roots and a considerable amount of ammonia removal is due to direct uptake by plants. However, the contribution of various hydroponic subsystem designs and plant species to water treatment in aquaponics systems has not been studied. Therefore, aquaponic system biofilters should be sized fairly close to the recommendations for recirculating systems.

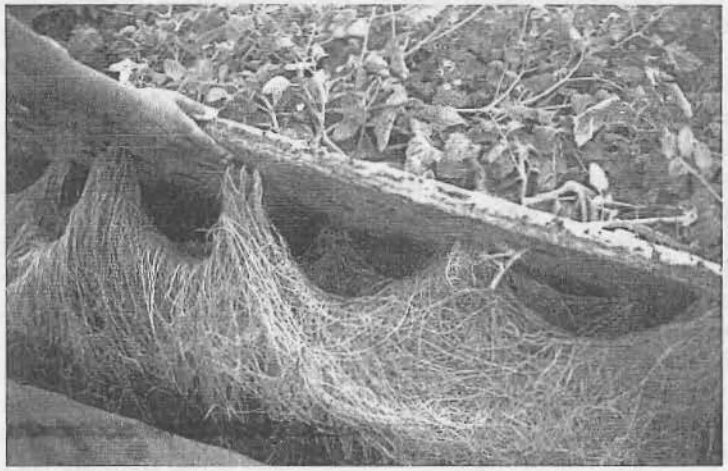
Nitrification efficiency is affected by pH. The optimum pH range for nitrification is 7.0 to 9.0, although most studies indicate that nitrification efficiency is greater at the higher end of this range. Most hydroponic plants grow best at pH in the range of 5.8 to 6.2. The acceptable range for hydroponic systems is 5.5 to 6.5. The pH of a solution affects nutrient solubility, especially trace metals. Essential nutrients such as iron, manganese, copper, zinc, and boron are less available to plants at pH above 7.0 while the solubility of phosphorus, calcium, magnesium, and molybdenum sharply decreases at pH below 6.0. Compromise between nitrification and nutrient availability is reached in aquaponic systems by maintaining pH close to 7.0.

Nitrification is most efficient when water is saturated with DO. The UVI commercial-scale system maintains DO levels near 80% saturation (6 to 7 mg/L) by aerating the hydroponic tanks with numerous small air diffusers, one every 1.2 m (4 ft) distributed along the long axis of the tanks. Reciprocating (ebb and flow) gravel systems expose nitrifying bacteria to high atmospheric oxygen levels during the dewatering phase. The thin film of water that flows through NFT channels absorbs oxygen by diffusion, but dense plant roots and associated organic matter can block water flow and create anaerobic zones, which precludes the growth of nitrifying bacteria and further necessitates the installation of a separate biofilter.

Ideally, aquaponic systems should be designed so that the hydroponic subsystem also serves as the biofilter, which eliminates the capital cost and operational expense of a separate biofilter. Granular hydroponic media such as gravel, sand, and perlite provide sufficient substrate for nitrifying bacteria and generally serve as the sole biofilter in some aquaponic systems, although they have a tendency to clog, as stated earlier. If serious clogging occurs due to organic matter overloading, gravel and sand filters can actually produce ammonia as organic matter decays, rather than remove it. If this occurs, the gravel or sand must be washed, and the system must be redesigned by installing a solids removal device prior to the granular biofilter, or the organic loading rate must be decreased by reducing fish stocking and feeding rates. McMurtry et al. (1990) relied on sand for hydroponic substrate and biofiltration while the early work at UVI (Rakocy, 1984) utilized gravel media for both functions. In some aquaponic systems, nitrification in the hydroponic component has supplemented that in the biofilter. Lewis et al. (1978) used pea gravel for hydroponic substrate in conjunction with an RBC for biofiltration. Watten and Busch (1984) used crushed-gravel hydroponic substrate and a trickling biofilter. When gravel was eliminated from UVI's experimental units, an RBC was installed for biofiltration. When the experimental units were scaled up the first time, two RBCs were installed.

A series of three stock management experiments were conducted in the scaled-up unit, in which the surface area of each RBC was 92.9 m^2 (1000 ft^2) and the total growing area for hydroponic lettuce was 71.4 m^2 (768 ft^2). In the first two experiments the feeding to the system ranged from 3 to 6 kg/day (6.6 to 13.2 lbs/day) and good water quality was maintained. A feeding rate of 4 kg/day (8.8 lbs/day) was nearly equivalent to the optimum design ratio (57 g of feed/day/m^2) for staggered lettuce production. In the third experiment the RBCs were eliminated to obtain a preliminary determination of the biofiltration

capacity of the hydroponic component. The maximum feeding rate to the system was increased to 8 kg/day (17.6 lbs/day), and water quality continued to be good. Average levels of ammonia-nitrogen and nitrite-nitrogen in the rearing tank were 1.3 and 0.7 mg/L, respectively. This experiment showed that a combination of direct ammonia uptake by lettuce plants and nitrification on the hydroponic tank floor, walls, and the underside of the floating polystyrene sheets can provide sufficient nitrification to maintain good water quality at a feeding rate that is two times greater than the optimum design ratio. The total surface area in each hydroponic tank was 92.9 m^2 (1000 ft^2), not including the additional surface area provided by the plant roots. The same surface area as contained in an RBC.



Massive root development of tomato plants supported by a sheet of polystyrene in UVI's Aquaponic System.

An experiment was conducted in UVI's replicated systems, to determine the waste treatment capacity of raft hydroponics (Gloger et al. 1995). Three identical systems were stocked with large *Oreochromis niloticus* (480 g/fish) so that feed could be incremented weekly until the waste treatment capacity was reached. The fish were initially underfed. Romaine lettuce (Parris Island) at a density of 29.6 plants/m^2 was produced continuously on a 4-week, staggered cycle. The results were based on the hydroponic growing area, expressed as g/m^2/day. The mean values and ranges were as follows: feeding rate, 159 g/m^2/day (77 to 230); wet weight plant production, 338 g/m^2/day; dry weight plant

production, 16.9 g/m^2/day; ammonia-nitrogen removal, 0.56 g/m^2/day (-1.23 to 2.29); nitrite-nitrogen removal, 0.62 g/m^2/day (-2.68 to 2.28); BOD removal, 4.78 g/m^2/day (0.0 to 4.92); COD removal, 30.29 g/m^2/day (-10.58 to 69.72); total nitrogen uptake by lettuce, 0.83 g/m^2/day; total phosphorus uptake by lettuce, 0.17 g/m^2/day. If the ammonia-nitrogen removal rate is divided by two to account for the surface area of the tank floor and the underside of the polystyrene sheet, the resultant value (0.28 g/m^2/day) falls within the range of removal rates reported for recirculating-system biofilters (Losordo, 1997). The maximum sustainable feeding rate was equivalent to 180 g/m^2/day, about three times greater than the optimum design ratio for the staggered production of Bibb lettuce in aquaponic systems.

Raft hydroponics not only provides adequate waste treatment for correctly-sized aquaponic systems and eliminates the need and expense of separate biofiltration units, but its excess treatment capacity ensures safe and stable water quality. After an initial acclimation period of about one month, experience with the UVI systems has shown that it is not necessary to monitor ammonia and nitrite values on a frequent basis, but weekly checks would be a prudent management practice.

Aquaponic systems using nutrient film technique (NFT) as the hydroponic component may require a separate biofilter. NFT consists of narrow plastic channels for plant support with a film of nutrient solution flowing through them. Compared to raft culture, the water volume and surface area of NFT are considerably smaller because there is just a thin film of water and no substantial side wall area and no raft underside surface area for colonization by nitrifying bacteria.

19.5 HYDROPONIC SUBSYSTEMS

A number of hydroponic subsystems have been used in aquaponics (Rakocy and Hargreaves, 1993). Gravel hydroponic subsystems are common in small operations. To ensure adequate aeration of plant roots, gravel beds have been operated in a reciprocating (ebb and flow) mode, where the beds are alternately flooded and drained (Lewis et al. 1978; Lewis et al. 1980; Sutton and Lewis, 1982; Wren, 1984; Rakocy, 1984), or in a dewatered state, in which culture water is applied continuously to the base of the individual plants through small-diameter plastic tubing (Watten and Busch, 1984). Depending on its composition, gravel can

provide some nutrients for plant growth, e.g., calcium is slowly released as the gravel reacts with acid produced during nitrification (Rakocy and Nair, 1987).

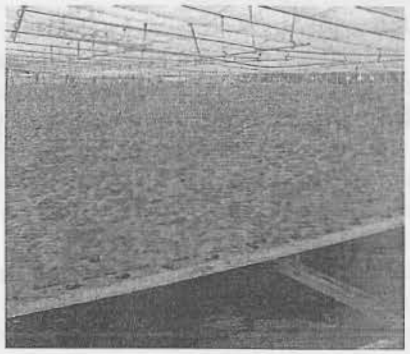
Gravel has several of negative aspects. The weight of gravel requires strong support structures. It is subject to clogging with suspended solids, microbial growth, and the roots that remain after harvest. The resulting reduction in water circulation together with decomposition of organic matter leads to the formation of anaerobic zones, which impairs or kills plant roots. The small plastic tubes used to irrigate gravel are also subject to clogging with biological growth. Moving and cleaning gravel substrate is difficult due to its weight. Planting in gravel is also difficult and plant stems can be damaged by abrasion in outdoor systems exposed to wind. Having high porosity, gravel retains very little water if drained. Disruption in flow will lead to the rapid onset of water stress (wilting). The sturdy infrastructure required supporting gravel and the potential of clogging imposes a size limitation on gravel beds.

One popular gravel-based aquaponic system uses pea gravel in 1.2 m x 2.4 m (4 ft x 8 ft) beds that are irrigated through a distribution system of PVC pipes over the gravel surface. Numerous small holes in the pipes distribute culture water on the flood cycles. The beds are allowed to drain completely between flood cycles. Solids are not removed from the culture water and organic matter does accumulate, but the beds are tilled between planting cycles, allowing some organic matter to be dislodged and discharged.

Sand has been used as hydroponic media in aquaponic systems (Ferguson, 1982; McMurtry et al. 1990). Ferguson (1982) raised 28 types of vegetables in trays of sand that were stacked three tiers high in a commercial greenhouse. The greenhouse produced an average of 8,700 kg of vegetables during 10-week growth cycles in trays (0.6–0.9 m wide) with a total length of 1,155 m. The trays were made of parachute cloth so that water applied to the upper tray slowly percolated through it to irrigate the trays beneath it. McMurtry et al. (1990) constructed hydroponic sand beds (7.5 m x 1.5 m x 0.5 m) on sloped ground that was covered by polyethylene sheets. The beds were adjacent to in-ground rearing tanks with their floors sloping to one side. A pump in the deep end of the rearing tank was activated for 30 minutes five times daily to furrow irrigate the adjacent sand bed. The culture water percolated

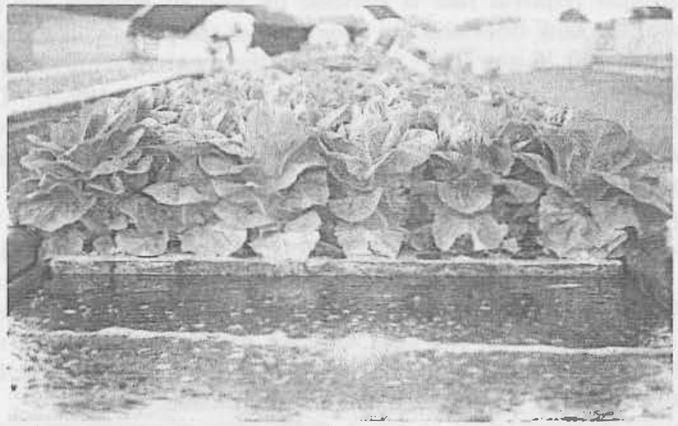
through the sand and returned to the rearing tank. The potential of sand substrates becoming clogged with solids can be reduced by regulating the solids loading rate. A coarse grade of sand is needed to reduce the potential for clogging over time and some solids should be removed prior to irrigation.

Perlite is another media that has been used in aquaponic systems. Perlite is placed in shallow aluminum trays 8 cm deep (3 inches) with a baked enamel finish. The trays vary from 20 cm to 1.2 m (8 inches to 4 ft) in width and can be fabricated to any length, but 6 m (20 ft) is the maximum recommended length. At intervals of 6 m (20 ft), adjoining trays should be separated by 8 cm (3 inches) or more in elevation so that effluent drops to the lower tray and becomes reaerated. A slope 1 to 144 (1 inch in 12 ft or 0.7%) is needed for water flow. A small trickle of water enters at the top of the tray, flows through the perlite, keeping it moist, and discharges into a trough at the lower end. Solids must be removed from the water before it enters the perlite tray. Full solids loading will clog the perlite, form short-circuiting channels, create anaerobic zones, and lead to non-uniform plant growth. Shallow perlite trays provide minimal area for root growth and are better for smaller plants such as lettuce and herbs.



Nutrient film technique (NFT) has been successfully incorporated into a number of aquaponic systems (Head, 1984; Burgoon and Baum, 1984). NFT consists of many narrow plastic troughs 10 to15 cm wide (4 to 6 inches) in which plant roots are exposed to a thin film of water that flows down the troughs, delivering water, nutrients, and oxygen to the roots of the plants. The troughs are lightweight, inexpensive, and versatile. Troughs can be mounted over rearing tanks to efficiently utilize vertical greenhouse space. However, this practice is discouraged if it interferes with fish and plant operations such as harvesting. High plant density can be maintained by adjusting the distance between troughs to provide optimum plant spacing during the growing cycle. Aquaponic systems utilizing NFT require effective solids removal to prevent excess solids accumulation on roots, which can lead to root death and poor plant growth. With NFT, a disruption in water flow can lead quickly to wilting and death. Water is delivered at one end of the troughs by a PVC manifold with discharge holes above each

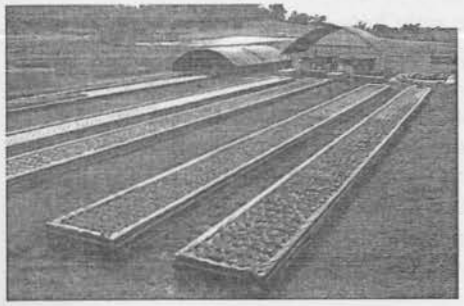
trough and collected at the opposite, down-slope end in an open channel or large PVC pipe. The use of mirotubes, which are used in commercial hydroponics, is not recommended because they will clog. The holes should be as large as practical to reduce cleaning frequency.



Raft culture of romaine lettuce in an aerated hydroponic tank in UVI's Aquaponic System.

A floating or raft hydroponic subsystem is ideal for the cultivation of leafy green and other types of vegetables (Zweig, 1986; Rakocy et al. 1989b). Long channels with closed-cell polystyrene sheets support vegetables at the water surface with roots suspended in the culture water (Jensen and Collins, 1985). The system provides maximum exposure of roots to the culture water and avoids clogging, although suspended solids captured by the roots can cause root death if concentrations are high (Zweig, 1986). The sheets shield the water from direct sunlight and maintain lower than ambient water temperatures. A disruption in pumping does not affect the plant's water supply as in gravel, sand, and NFT subsystems. The sheets are easily moved along the channel to a harvesting point where they can be lifted out of the water and placed on supports at an elevation that is comfortable for the workers.

The UVI system uses three sets of two raft hydroponic tanks that are 30.5 m (100 ft) long by 1.22 m (4 ft) wide by 40.6 cm (16 inches) deep and contain 30.5 cm (12 inches) of water. The channels are lined with low density polyethylene liners (20 mils thick) and covered by expanded polystyrene sheets (rafts), which are 2.44 m (8 ft) long by 1.22 m (4 ft) wide by 3.8 cm (1.5 inches) thick. Net pots are placed in holes in the raft



and just touch the water surface. Two inch net pots are generally used for leafy green plants while 7.62 cm (3-inch) net pots are used for larger plants such as tomatoes or okra. Holes of the same size are cut into the polystyrene sheet. A lip at the top of the net pot secures the net pot and keeps it from falling through the hole into the water. Seedlings are nursed in a greenhouse and then placed into net pots, and their roots grow into the culture water while their canopy grows above the raft surface.

A disadvantage of rafts in an aquaponic system is that roots are exposed to harmful organisms associated with aquaculture systems. For example, if tilapia fry gain access to the raft tanks, they consume plant roots and thereby severely stunt growth, although it is relatively easy to prevent the entry of tilapia by using a fine mesh screen. Similarly, blooms of zooplankton, especially ostracods, will consume root hairs and fine roots, retarding plant growth. Other pests are tadpoles, snails, and leeches that consume roots and nitrifying bacteria. These problems are surmounted by stocking some carnivorous fish that prey on pests in the hydroponic tanks. At UVI, snails are controlled with shellcracker sunfish (*Lepomis microlophus*), and zooplankton are controlled with black tetra (*Gymnocorymbus ternetzi*).

19.6 Sump

Water flows by gravity from gravel, sand, and raft hydroponic subsystems to a sump, which is the lowest point in the system. The sump contains a pump or pump inlet which returns the treated culture water to the rearing tanks. If NFT troughs or perlite trays are located above the rearing tanks, the sump would be positioned in front of them so that water could be pumped up to the hydroponic component for gravity return to the rearing tanks. There should be only one pump to circulate water in an aquaponic system.

The sump should be the only tank in the system where the water level decreases as a result of overall water loss from evaporation, transpiration, sludge removal, and splashing. A mechanical valve is used for the automatic addition of replacement water from a storage reservoir or well. Municipal water should not be used unless it is de-chlorinated and surface water should not be used because it may contain disease organisms. A water meter should be used to record additions. Unusually high water consumption indicates a leak.

The sump is a good location for the addition of base to the system. Soluble base such as potassium hydroxide causes high and toxic pH levels in the sump. However, as water is pumped into the rearing tank, it is diluted and pH decreases to acceptable levels. The UVI system uses a separate base addition tank located next to the sump. As water is pumped from the sump to the fish rearing tanks, a small pipe, tapped into the main water distribution line, delivers a small flow of water to the base addition tank, which is well aerated with one large air diffuser. Base is added to this tank as needed to maintain a pH of 7.0 in the system. The base dissolves, gradually enters the sump and is pumped to the rearing tanks where it is quickly diluted in large volumes of turbulent water. Gradual addition of base avoids spikes in pH values, which are harmful to both fish and plants.

19.7 Construction Materials

A wide range of materials are used to construct aquaponic systems. Budget limitations often play a major role in selecting inexpensive and questionable materials such as vinyl-lined, steel-walled swimming pools. Plasticizers used in vinyl manufacture are toxic to fish. The liners must be washed thoroughly or aged with water for several weeks before fish can be added safely to a tank of clean water. After a few growing periods, vinyl liners shrink upon drying, become brittle and crack, while the steel walls gradually rust. Nylon-reinforced, neoprene-rubber liners are not recommended either. Tilapia eat holes in rubber liners at the folds by grazing on microorganisms. Moreover, neoprene-rubber liners are not impervious to chemicals. If herbicides and soil sterilants are applied under or near rubber liners, these chemicals can diffuse into culture water, accumulate in fish tissue, and kill hydroponic vegetables.

Wood is not considered to be a good construction material for aquaponic systems because it prone to rotting in the high humidity environment. If wood is used, it must be untreated as treated lumber contains toxic compounds such as arsenic to inhibit bacterial growth. If

these compounds leach into the water, they could affect the beneficial bacteria that the system depends on and contaminate the fish and vegetables. Untreated wood must be waterproofed with fiberglass matt and resin on the inside and epoxy paint on the outside. Wooden tanks must not be in contact with soil to prevent the entry of termites. In general, wooden tanks have a short life span.

Fiberglass is the best construction material for the rearing tanks, sump, and filter tanks. Fiberglass tanks are sturdy, durable, non-toxic, movable, and easy to plumb. An alternative to fiberglass is concrete, which is cheaper in many countries, although it lacks the flexibility of fiberglass construction. Commercially available NFT troughs, made from extruded polyethylene, are specifically designed to prevent puddling and water stagnation leading to root death and are preferable to makeshift structures (rain gutters, PVC pipes, etc.). Plastic troughs are commercially available for floating hydroponic subsystems, but they are expensive. A suitable alternative is to use of polyethylene liners and concrete-block or poured-concrete walls. Four types of liners have been tested in UVI's commercial-scale system. They are high-density polyethylene [1.5 mm (60 mil) and 0.5 mm (20 mil)], low-density polyethylene [0.5 mm (20 mil)], and a thick grade of nylon-reinforced vinyl with an under layer of high-density polyethylene. All of these liners are performing well after 5 to10 years, but it appears that 0.5-mm high-density polyethylene liners (HDPE) are best. They are easy to install, relatively inexpensive and durable, having an expected service life of 12 to 15 years. Initially, HDPE liners were black, but recently UV-resistant white liners have been introduced. White liners are preferable in that they reflect light and do not become as hot as black liners. This is an important characteristic in the tropics and during summers in temperate climates where the goal is to avoid high water temperatures.

19.8 Component Ratios

Aquaponic systems are generally designed to meet the size requirements for solids removal (for those systems requiring solids removal) and biofiltration (if a separate biofilter is used) for the amount of fish being raised. After the size requirements are calculated, it is prudent to add excess capacity as a safety margin. However, if a separate biofilter is used, the hydroponic component is the safety factor because a significant amount of ammonia uptake and nitrification will occur regardless of hydroponic technique.

Another key design criterion is the ratio between the fish rearing and hydroponic components. The key aspect of the criterion is the ratio of daily feed input to plant growing area. If the ratio of daily feeding rate to plant growing area is too high, nutrient salts will accumulate rapidly and may reach phytotoxic levels. Higher water exchange rates will be required to prevent excessive nutrient buildup. If the ratio of daily feeding rate to plants is too low, plants will develop nutrient deficiencies and more nutrient supplementation will be required. Fortunately, hydroponic plants grow well over a wide range of nutrient concentrations.

The optimum ratio of daily fish feed input to plant growing area will maximize plant production while maintaining relatively stable levels of dissolved nutrients. A volume ratio of 1 m^3 of fish rearing tank to 2 m^3 of pea gravel 3 to 6-cm (1/8 to 1/4 inch) in diameter as hydroponic media is recommended for reciprocating (flood and drain) gravel aquaponic systems. This ratio requires that tilapia are raised to a final density of 60 kg/m^3 (0.5 lb/gallon) and fed appropriately. With the recommended ratio no solids are removed from the system. The hydroponic beds should be cultivated (stirred up) between crops and inoculated with red worms to help break down and assimilate the organic matter. With this system nutrient supplementation may not be necessary.

"Rule of Thumb"

Pea Gravel Hydroponic Media
1 m^3 of fish tank volume to 2 m^3 of hydroponic media

As a general guide for raft aquaponics, a ratio in the range of 60-100 g of fish feed/m^2 of plant growing area per day should be used. Ratios within this range have been used successfully in the UVI system for the production of tilapia, lettuce, basil and several other plants. In the UVI system all solids are removed, with a residence time of <1 day for settleable solids (>100 microns) removed by a clarifier, and 3 to 4 days for suspended solids removed by an orchard netting filter. The system uses rainwater, and supplementation is required for potassium, calcium, and iron.

"Rule of Thumb"

60-100 g of fish feed per day per square meter of plant growing area for the staggered production of leaf lettuce.

Another factor to consider in determining the optimum feeding rate ratio is the total water volume of the system, which affects nutrient concentrations. In raft hydroponics, approximately 75% of the system water volume is in the hydroponic component whereas gravel beds and NFT troughs contain minor amounts of system water. Theoretically in systems producing the same quantity of fish and plants, a daily feeding rate of 100 g/m^2 for example would produce total nutrient concentrations nearly four times higher in gravel and NFT systems (e.g., 1,600 mg/L) as compared to raft systems (e.g., 400 mg/L), but total nutrient mass would be equal among systems. Nutrient concentrations outside acceptable ranges affect plant growth. Therefore, the optimum design ratio varies depending on the type of hydroponic component. Gravel and NFT systems should have a feeding rate ratio that is approximately 25% of the recommended ratio for raft hydroponics.

Other factors involved in determining the optimum feeding rate ratio are the water exchange rate, nutrient levels in the source water, degree, and speed of solids removal and type of plant being raised. Lower rates of water exchange, higher source-water nutrient levels, incomplete or slow solids removal, resulting in the release of more dissolved nutrients through mineralization, and slower growing plants would allow a lower feeding rate ratio. Conversely, higher water exchange rates, low source-water nutrient levels, rapid and complete solids removal and fast growing plants would allow a higher feeding rate ratio.

The optimum feeding rate ratio is influenced by the plant culture method. With batch culture all plants in the system are planted and harvested at the same time. During their maximum growth phase, there is a large uptake of nutrients, which requires a higher feeding rate ratio during that period. In practice, however, a higher feeding rate ratio is used throughout the production cycle. With a staggered production system, plants are in different stages of growth, which levels out nutrient uptake rates and allows good production with slightly lower feeding rate ratios.

In properly designed aquaponic systems, the surface area of the hydroponic component is quite large compared to the surface area of the fish rearing tanks if it is stocked at commercial levels. The commercial-scale unit at UVI has a ratio of 7.3:1. The total plant growing area is 214 m^2 (2304 ft^2) compared to total fish rearing surface area of 29.2 m^2 (314 ft^2). With diffused aeration, final tilapia densities have reached a mean of 76 kg/m^3 (0.63 lbs/gal) With pure oxygen, final densities can be increased to 120 kg/m^3 (1.0 lb/gal) or greater. Therefore, smaller rearing tanks could be used to produce the same amount of fish and a ratio of 11.5:1 would be more appropriate.

"Rule of Thumb"
11.5 to 1 ratio of plant beds to fish tank surface area in high density fish systems (120 kg/m^3)

19.9 Plant Growth Requirements

Maximum plant growth in aquaponic systems requires 16 essential elements for proper nutrition. These nutrients are referred to below, in the order of their concentrations (mg/L) in plant tissue with carbon and oxygen being the highest. The essential elements are arbitrarily divided into macronutrients, those required in relatively large quantities, and micronutrients, those required in considerably smaller amounts. Three of the macronutrients, carbon (C), oxygen (O) and hydrogen (H), are supplied by water (H_2O) and carbon dioxide gas (CO_2). All of the remaining nutrients are absorbed from the culture water. Other macronutrients include nitrogen (N), potassium (K), calcium (Ca), magnesium (Mg), phosphorus (P), and sulfur (S). The seven micronutrients include chlorine (Cl), iron (Fe), manganese (Mn), boron (B), zinc (Zn), copper (Cu), and molybdenum (Mo). All of these nutrients must be in proper balance for optimum plant growth. High levels of one nutrient can influence the bioavailability of others. For example, excessive amounts of potassium may interfere with uptake of magnesium or calcium while excessive amounts of either of the latter nutrients may interfere with the uptake of the other two nutrients (Gerber, 1985).

An elevated CO_2 level in the atmospheric environment of unventilated hydroponic structures has given dramatic increases in crop yield in northern latitudes (Jensen and Collins, 1985). Kimball (1982) summarized the data on CO_2 enrichment from more than 360 observations on 24 crops in a series of 50 reports. The study showed that doubling atmospheric CO_2 increased agricultural yields by an average of 30%. The high cost of energy to generate CO_2 has discouraged its use in conventional hydroponic systems. However, an enclosed aquaponic system is ideal for generating CO_2 due to the huge amounts that are constantly vented from the culture water.

Luther (1990, 1991A, 1991B, 1991C, 1993) cites a growing body of evidence that healthy plant development relies on a wide range of organic compounds in the root environment. These compounds, generated by complex biological processes involving microbial

decomposition of organic matter, include vitamins, auxins, gibberellins, antibiotics, enzymes, coenzymes, amino acids, organic acids, hormones, and other metabolites. These compounds are directly absorbed and assimilated by the plants and stimulate growth, enhance yields, increase vitamin, and mineral content, improve fruit flavor and hinder the development of pathogens. Various fractions of dissolved organic matter, e.g., humic acid, form organo-metalic complexes with Fe, Zn, and Mn, thereby increasing the availability of these micronutrients to plants (Chen and Solvitch, 1988). Luther states that although inorganic nutrients are necessary for plant survival, plants need organic metabolites from the environment to reach full hereditary potential.

Maintaining high DO levels in the culture water is extremely important for optimal plant growth, especially in aquaponic systems with their high organic loads. Hydroponic plants are subject to intense root respiration and they draw large amounts of oxygen from the surrounding water. If DO is deficient, root respiration decreases, resulting in reduced water absorption, decreased nutrient absorption, loss of cell tissue from roots and a reduction in plant growth (De Wit, 1978). Low DO levels correspond with high concentrations of carbon dioxide, conditions that promote the development of plant root pathogens. Chun and Takakura (1993) tested four nutrient-solution DO levels for hydroponic lettuce and found that root respiration, root growth, and transpiration were greatest at saturated DO levels.

Climatic factors also influence hydroponic vegetable production. Production is generally best in regions with maximum intensity and duration of light. Jensen and Collins (1985) reported that growth rates of lettuce plants in an Arizona greenhouse correlated positively with levels of available light up to the highest levels measured, although radiation levels in the Arizona desert are two to three times that of more temperate climates. When 30% shade cloth was used to cover lettuce plants in the UVI system, also in a region of intensive solar radiation, the plants elongated, the leaves twisted around the stem, and production declined. Growth slows substantially in temperate greenhouses during winter due to low solar radiation. Supplemental illumination can improve wintertime production, but it is not generally cost effective.

Water temperature is far more important than air temperature for hydroponic plant production. The best water temperature for most hydroponic crops is around is 20–22°C (68-75 °F). However, water temperature can go as low as the mid-60s for most common garden crops and slightly lower for winter crops such as cabbage, brussel sprouts and broccoli. Maintaining the best water temperature requires heating during the winter in temperate greenhouses and year-round cooling in tropical

greenhouses. In addition to evaporative cooling of tropical greenhouses, chillers are often used to cool the nutrient solution. In tropical outdoor systems, complete shading of the fish rearing and filtration components lowers system water temperature. In raft hydroponics, the polystyrene sheets shield water from direct sunlight and maintain temperatures that are several degrees lower than those in open water bodies. Seasonal adjustment in selection of plant crop varieties may be necessary for both temperate and tropical aquaponic production. Plants cultured in outdoor aquaponic systems must be protected from strong winds, especially following transplanting when seedlings are fragile on most vulnerable to damage.

19.10 Nutrient Dynamics

Collectively dissolved nutrients are measured as total dissolved solids (TDS), expressed as mg/L, or as the capacity of the nutrient solution to conduct an electrical current (EC), expressed as millimhos/cm (mMho/cm). In a hydroponic solution the recommended range for TDS is 1,000 to 1,500 (1.5 to 3.5 mMho/cm). In an aquaponic system considerably lower levels of TDS (200 to 400 mg/L) or EC (0.3 to 0.6 mMho) will produce good results because nutrients are generated continuously. A concern with aquaponic systems is nutrient accumulation. High feeding rates, low water exchange, and insufficient plant growing areas can lead to the rapid buildup of dissolved nutrients to potentially phytotoxic levels. Phytotoxicity is encountered at TDS concentrations above 2,000 mg/L or EC above 3.5 mMho. Since aquaponic systems are characterized by variable environmental conditions such as daily feed input, solids retention, mineralization, water exchange, nutrient input from source water or supplementation, and variable nutrient uptake by different plant species, it is difficult to predict the exact level of TDS or EC and how it is changing. Therefore, the culturist should purchase an inexpensive conductivity meter and periodically measure TDS or EC. If dissolved nutrients are steadily increasing and approaching 2,000 mg/L as TDS or 3.5 mMho as EC, increasing the water exchange rate or reducing the fish stocking rate and feed input will quickly reduce nutrient accumulation. Since these methods either increase costs (i.e., more water consumed) or lower output (i.e., less fish produced), they are not good long-term solutions. Better but more costly solutions involve increased solids removal (i.e., upgrade the solids removal component) or enlarged plant growing areas.

Early work with UVI's experimental systems showed that conductivity measurements of TDS increased steadily as increasing quantities of feed were added to the system (Rakocy et al. 1993). Phytotoxic levels were reached after the addition of approximately 10 kg feed/m^3 of system volume. In an experiment to determine the optimum ratio of daily feed input to leaf lettuce growing area, the concentration of TDS increased by 147.5 g/kg of dry weight of feed at the optimum ratio of 57 g/day/m^2. However, during the first 8 months of operation of an early model of UVI's commercial-scale system, 26.9 kg of feed/m^3 of system volume was applied and the highest conductivity level was only 890 mg/L as TDS. The accumulation rate for TDS was 26 g/kg of dry weight of feed, Table 19.3. Three factors contributed to the lower nutrient-accumulation rate. The actual mean ratio of daily feed input to plant growing area (49.5 g/day/m^2) turned out to be less than the optimum design ratio. Lettuce productivity was greater in the commercial-scale system. Romaine and leaf lettuce plants grew to a size of 250–650 g in 4 weeks (8.9–23.2 g/day) in the commercial-scale system compared to an average Bibb lettuce size of 131 g in three weeks (6.2 g/day) in the experimental system. The average reduction of conductivity on passage through the hydroponic component during the first 8 months was 4.2 mg/L as TDS. Substantial amounts of solids were removed by the filter tanks and consequently less mineralization may have occurred than in the experimental systems.

Table 19.3 Nutrient Accumulation (g/kg Dry Weight Feed) of Conductivity and Major Cations and Anions from Two Experimental Aquaponic Systems and a Commercial-Scale Aquaponic System Using Raft Hydroponics for Lettuce Production

Nutrient	Exp. System 1[a]	Exp. System 2[b]	Commercial System[c]
Conductivity[d]	215.2	147.5	26.2
NO_3-N	35.6	14.9	3.7
PO_4-P	3.0	-	0.2
SO_4-S	1.9	1.8	0.6
K	66.0	-	4.2
Ca	-	7.3	2.3
Mg	1.2	1.8	0.4

[a] Supplementation with K but not Ca. Minor, one-time supplementation with P. From Rakocy et al. 1993

[b] Optimum feed to growing area ratio (57 g/day/m^2) from ratio study (Rakocy et al. 1993). K and Ca supplementation

[c] From the first 8 months of operation of an early model of the commercial-scale system. K and Ca supplementation

[d] As TDS

The major ions that contribute to increased conductivity are nitrate (NO_3^-), phosphate (PO_4^{-2}), sulfate (SO_4^{-2}), K^+, Ca^{+2} and Mg^{+2}. Levels of NO_3^-, PO_4^{-2} and SO_4^{-2} are usually sufficient for good plant growth while levels of K^+ and Ca^{+2} are generally insufficient for maximum plant growth. Potassium is added to the system in the form of potassium hydroxide (KOH) while Ca is added as calcium hydroxide [$Ca(OH)_2$]. In some systems Mg may be limiting. In the UVI commercial-scale system, KOH and $Ca(OH)_2$ are added in equal amounts usually in the range of 500-1,000 g in the UVI system. The bases are added alternately several times weekly to maintain pH near 7.0. Adding basic compounds of K and Ca serves the dual purpose of supplementing essential nutrients and neutralizing acid. Magnesium can be supplemented by using dolomite [$CaMg(CO_3)_2$] as the base to adjust pH. The addition of too much Ca can lead to the precipitation of phosphorous from culture water in the form of dicalcium phosphate [$CaHPO_4$]. All of the macronutrients with the exception of orthophosphate have a strong correlation with feed input (Rakocy et al. 1993).

The accumulation of nitrate ions is a concern with aquaponic systems. The discharge from one experimental system at UVI contained 180 mg/L as NO_3-N (Rakocy, 1994). The installation of the filter tanks in the UVI commercial-scale system provided a mechanism for controlling nitrate levels through the process of denitrification, the reduction of nitrate ions to nitrogen gas by anaerobic bacteria. Large quantities of organic matter accumulate on the orchard netting between cleanings. Denitrification occurs in anaerobic pockets that develop in the sludge. The entire water column moves through the accumulated sludge, which provides good contact between nitrate ions and denitrifying bacteria. The frequency of cleaning the netting regulates the degree of denitrification. When the netting is cleaned frequently, e.g., twice per week, sludge accumulation and denitrification is minimized, which leads to an increase in nitrate concentrations. When the netting is cleaned less frequently, e.g., once per week, sludge accumulation and denitrification are maximized, which leads to a decrease in nitrate levels. Nitrate-nitrogen levels can be regulated within a range of 1 to 100 mg/L or higher. High nitrate concentrations promote the growth of leafy green vegetables while low nitrate concentrations promote fruit development in vegetables such as tomatoes.

Denitrification recovers the alkalinity that is lost in the nitrification process. Sometimes an aquaponic system will go for long periods of time with no change in pH without any need to add bases such as calcium hydroxide or potassium hydroxide. Stable pH in an aquaponic system indicates that too much denitrification is occurring (somewhat counter intuitive). If there is no need to add base, the plants could develop calcium and potassium deficiencies. When pH is stable, the frequency of cleaning the filter tanks should be increased and any anaerobic zones that have been created due to the accumulation of solids in the system should be removed.

In a study using raft hydroponics for lettuce production, Seawright (1995) obtained similar results for macronutrient accumulation with two important exceptions: P accumulated in relation to feed input, but there was no significant relationship between feed input and N. Sodium bicarbonate ($NaHCO_3$) was added for pH control. The addition of Ca bases in the UVI system may have contributed to the precipitation of P from the culture water in the form of calcium phosphate [$Ca_3(PO_4)_2$]. Seawright removed solid waste from the system once per week and therefore denitrification may have been greater than in the UVI system where solid waste is removed at least twice per day from the clarifier and up to twice per week from the filter tanks. Although Seawright did not supplement with K, it accumulated with respect to feed input. However, plants require high levels of K and supplementation is needed in aquaponic systems. Seawright's finding that Ca is negatively correlated with feed input agrees with early work at UVI (Rakocy and Nair, 1987). Some investigators have found that Mg is limiting (Pierce, 1980; Head, 1984; Zweig, 1986). Magnesium can be supplemented by using dolomite [$CaMg(CO_3)_2$] as the base to adjust pH.

Sodium bicarbonate ($NaHCO_3$) should never be added to an aquaponic system for pH control. The accumulation of Na is a concern in aquaponic systems because high Na levels in the presence of chloride are toxic to plants (Resh, 1995). The maximum Na concentration in hydroponic nutrient solutions should not exceed 50 mg/L (Verwer and Wellman, 1980). Higher Na levels will interfere with the uptake of K and Ca (Douglas, 1985). In lettuce, reduced Ca uptake leads to tip burn, resulting in an unmarketable plant (Collier and Tibbitts, 1982). In UVI's systems, tip burn has occurred during the warmer months. Soluble salt (NaCl) levels in fish feed are relatively high. In the initial commercial-scale system, Na reached 51.0 mg/L in the 6th month and then declined to 37.8 mg/L in the 8th month, possibly due to rainfall dilution. The Na accumulation rate through the 6th month was 2.56 g/kg of dry weight feed. If Na exceeds 50 mg/L and the plants appear to be affected, a

partial water exchange (dilution) may be necessary. Rainwater is used in UVI's systems because the groundwater of semiarid islands generally contains too much salt for aquaponics.

Table 19.4 Mean Concentration (mg/L) of Micronutrients from Two Experimental Aquaponic Systems and a Commercial-Scale Aquaponic System Using Raft Hydroponics for Lettuce Production

	Micronutrient					
System	Fe	Mn	Cu	Zn	B	Mo
Exp. 1[a]	1.79	0.04	0.12	0.78	0.16	-
Exp. 2[b]	0.48	0.13	0.07	0.68	-	-
Commercial[c]	0.57	0.05	0.05	0.44	0.06	0.006
HNF[d]	5.0	0.5	0.03	0.05	0.5	0.02
HNF[e]	5.0	0.5	0.1	0.1	0.5	0.05

[a] From Rakocy et al. 1993.
[b] Optimum feed to growing area ratio (57 g/day/m^2) from ratio study (Rakocy et al. 1993).
[c] From the first 8 months of operation of a commercial-scale system.
[d] Hydroponic nutrient formulation for lettuce grown in the tropics (Resh, 1995).
[e] Hydroponic nutrient formulation for lettuce grown in Florida and California (Resh, 1995).

With the exception of Zn^{+2}, the micronutrients Fe^{+2}, Mn^{+2}, Cu^{+2}, B^{+3} and Mo^{+6} do not accumulate significantly in aquaponic systems with respect to cumulative feed input, Table 19.4 (Rakocy et al. 1993). The Fe^{+2} derived from fish feed is insufficient for hydroponic vegetable production and must be supplemented (Lewis et al. 1980; MacKay and Van Toever, 1981; Zweig, 1986). Chelated Fe^{+2} should be applied at a rate to achieve a Fe^{+2} concentration of 2.0 mg/L. Chelated Fe^{+2} has an organic compound attached to the metal ion to prevent it from precipitating out of solution and making it unavailable for plant nutrition. The best chelate is Fe-DTPA because it remains soluble at pH 7.0. Fe-EDTA is commonly used in the hydroponics industry, but it is less stable at pH 7.0 and needs to be replenished frequently. Fe^{+2} may also be applied in a foliar spray from which Fe^{+2} is absorbed directly through plant leaves. A comparison of Mn^{+2}, B^{+3} and Mo^{+6} levels with standard nutrient formulations for lettuce shows that their concentrations in

aquaponic systems are several times lower than their initial levels in hydroponic formulations. Deficiency symptoms for Mn^{+2}, B^{+3} and Mo^{+6} are not detected in aquaponic systems, and so their concentrations appear to be adequate for normal plant growth. Concentrations of Cu^{+2} are similar in aquaponic systems and hydroponic formulations while Zn^{+2} accumulates in aquaponic systems to levels that are four to sixteen times higher than initial levels in hydroponic formulations. Nevertheless, Zn^{+2} concentrations usually remain within the upper limit for fish safety which is 1 mg/L in hydroponic solutions (Douglas, 1985).

Seawright (1995) worked on the development of a "designer diet" for aquaponic systems that would generate nutrients in proportion to their requirements for hydroponic plant nutrition, thereby creating stable and balanced nutrient concentrations over prolonged periods. Data on the change in nutrient concentrations in relation to dietary nutrient input was collected for the co-culture of *O. niloticus* and romaine lettuce (Jericho). This data was used to develop a mass balance model theoretically capable of predicting the nutrient inclusion rates required in fish diets to maintain stable dissolved nutrient concentrations in aquaponic systems. The model was validated by applying a specially-formulated "designer diet" to an aquaponic system and maintaining near-equilibrium concentrations of Ca, K, Mg, N and P, suitable concentrations of Mn and Cu and acceptable accumulation rates of Na and Zn. The results showed that Cu, Fe, and Mn are not good candidates for dietary manipulation because of low bioavailability. Phosphorus is a good candidate at sub-neutral pH, but it precipitates from solution at basic pH. Sulfur, B and Mo were not tested. The fish grew well on the diet, but the development of bacterial diseases indicated that elevated nutrient levels may have lowered their disease resistance.

19.11 VEGETABLE SELECTION

Many types of vegetables have been grown in aquaponic systems, Tables 19.5-19.7. However, the goal is to select a vegetable for culture that will generate the highest level of income per unit area per unit time. Using this criterion, culinary herbs are the best choice. They grow very rapidly and command high market prices. The income from herbs such as basil, cilantro, chives, parsley, portulaca, and mint are many times higher than that from fruiting crops such as tomatoes, cucumbers, eggplant, and okra. For example, in experiments in UVI's commercial-scale system, basil production was 11,000 lbs annually at a value of $110,000 compared to annual production of 6,400 lbs of okra at a value of $6,400.

Fruiting crops also require longer culture periods (90 days or more) and are subject to more pest problems and diseases. Lettuce is another good crop for aquaponic systems because it can be produced in a short period (3 to 4 weeks in the system), and, as a consequence, pest pressure is relatively low. Unlike fruiting crops, a high proportion of the harvested biomass is edible. Other suitable crops include Swiss chard, pak choi, Chinese cabbage, collard and watercress. The cultivation of flowers has potential in aquaponic systems. Good results have been obtained with marigold and zinnia in UVI's aquaponic system. Traditional medicinal plants and plants used for the extraction of modern pharmaceuticals have not been cultivated in aquaponic systems, but there may be potential in growing some of these plants.

Table 19.5 Varieties and Yields of Cucumbers Evaluated in Aquaponic Systems[a]. Environmental Codes. TEMP=Temperate Zone, TROP=Tropical Zone, O=Outside, GH=Greenhouse

Variety	Yield (kg/plant)	Environment	Reference
Triumph	4.1	TEMP/O	Lewis et al. 1980
Patio Pik	1.6	TEMP/O	Lewis et al. 1980
Corona (Stokes)	28.6[b]	TEMP/GH	Burgoon and Baum, 1984
Bruinsma	8.2[b]	TEMP/GH	Burgoon and Baum, 1984
Vetomil			
Superator	4.1	TEMP/GH	Head, 1984
Sprint 4405	0.7	TEMP/GH	Wren, 1984
Burpee Hybrid 11	7.3[b]	TEMP/GH	McMurtry, 1990

[a] From Rakocy and Hargreaves, 1993
[b] kg/m^2

Table 19.6 Varieties and Yields of Leafy Greens (Lettuce, Pak Choi, Chinese Cabbage, Spinach) Evaluated in Aquaponic Systems[a]. Environment Codes. TEMP=Temperate Zone, TROP=Tropical Zone, O=Outside, GH=Greenhouse

Variety	Yield (g/plant)	Environment	Reference
Ostinata	162	TEMP/GH	Baum, 1981
Reskia	236	TEMP/GH	Burgoon and Baum, 1984
All Year Round	236	TEMP/GH	Burgoon and Baum, 1984
Karma	98	TEMP/GH	Burgoon and Baum, 1984
Ravel	45–50	TEMP/GH	Burgoon and Baum, 1984
Salina	44	TEMP/GH	Burgoon and Baum, 1984
El Captain	43	TEMP/GH	Burgoon and Baum, 1984
Bruinsma Columbus	0	TEMP/GH	Burgoon and Baum, 1984
Salad Bowl	-	TEMP/GH	Head, 1984
Pak Choi	-	TEMP/GH	Head, 1984
Winter Bloomsdale (Spinach)	-	TEMP/GH	Head, 1984
Buttercrunch	-	TEMP/GH	Zweig, 1986
Buttercrunch	193	TROP/O	Rakocy, 1989B
Summer Bibb	180	TROP/O	Rakocy, 1989B
Le Choi	508	TROP/O	Rakocy, 1989B
Pak Choi	422	TROP/O	Rakocy, 1989B
50-Day Hybrid (Chinese Cabbage)	638	TROP/O	Rakocy, 1989B
Tropical Delight (Chinese Cabbage)	589	TROP/O	Rakocy, 1989B
SummerBibb	107–116	TEMP/GH	Parker et al. 1990
Sierra	182–340	TROP/O	Rakocy et al. 1997
Nevada	149–360	TROP/O	Rakocy et al. 1997
Jerhico	267–344	TROP/O	Rakocy et al. 1997
Parris Island	181–446	TROP/O	Rakocy et al. 1997

[a] From Rakocy and Hargreaves (1993) and some more recent data.

Table 19.7 Varieties and Yields of Tomatoes Evaluated in Aquaponic Systems[a]. Environmental Codes: TEMP=Temperate Zone, TROP=Tropical Zone, O=Outside, GH=Greenhouse

Variety	Yield (kg/plant)	Environment	Reference
Tropic	0	TEMP/GH	Landesman, 1977
Floradel	5.4	TEMP/O	Lewis et al. 1978
Campbells 1327	4.6	TEMP/O	Lewis et al. 1978
San Marzano	4.6	TEMP/O	Lewis et al. 1978
Sweet 100	6.2	TEMP/O	Lewis et al. 1980
Better Boy	4.5	TEMP/O	Lewis et al. 1980
Rampo	3.9	TEMP/O	Lewis et al. 1980
Campbells 1327	2.3	TEMP/O	Lewis et al. 1980
Sweet 100	1.9	TEMP/GH	Baum, 1981
Spring Set	1.6	TEMP/GH	Baum, 1981
Sweet 100	0	TEMP/GH	MacKay et al. 1981
Jumbo	0	TEMP/GH	MacKay et al. 1981
Michigan Ohio Forcing	0	TEMP/GH	MacKay et al. 1981
Burpee Big Boy	0.2	TEMP/O	Markin, 1982
Floradel	8.9, 9.1	TEMP/O	Sutton and Lewis, 1982
Korala #127	6.8[b]	TEMP/GH	Burgoon and Baum, 1984
Vendor	4.1	TEMP/GH	Head, 1984
Tropic	0.5, 3.2	TROP/O	Watten and Busch, 1984
Homestead	2.7	TROP/O	Watten and Busch, 1984
Red Cherry	1.5	TROP/O	Watten and Busch, 1984
Prime Beefsteak	1.4	TROP/O	Watten and Busch, 1984
Vendor	0	TROP/O	Nair et al. 1985
Tropic	0	TROP/O	Nair et al. 1985
Jumbo	0	TROP/O	Nair et al. 1985
Perfecta	0	TROP/O	Nair et al. 1985
Laura	2.3–3.4	TEMP/GH	McMurtry, 1989
Kewalo	2.5–5.0	TEMP/GH	McMurtry, 1989
Sunny	10.1	TROP/O	Rakocy, 1989
Floradade	9.0	TROP/O	Rakocy, 1989
Vendor	3.7	TROP/O	Rakocy, 1989
Cherry Challenger	2.9	TROP/O	Rakocy, 1989
Champion	4.6[b]	TEMP/GH	McMurtry et al. 1990

[a] From Rakocy and Hargreaves, 1993
[b] kg/m^2

19.12 Crop Production Systems

There are three strategies for producing vegetable crops in the hydroponic component. These are staggered cropping, batch cropping, and intercropping. A staggered crop production system is one in which groups of plants in different stages of growth are cultivated simultaneously in the hydroponic subsystem. This production system allows regular harvest of produce and relatively constant uptake of nutrients from the culture water. This system is most effectively implemented where crops can be grown continuously, as in the tropics, subtropics, or temperate greenhouses with environmental control (Zweig, 1986). At UVI, the production of leaf lettuce is staggered so that a crop can be harvested weekly on the same day, which facilitates marketing arrangements. Bibb lettuce reaches market size in three weeks from transplanting. Therefore, three growth stages of Bibb lettuce are cultivated simultaneously, and one third of the crop is harvested weekly. Red leaf lettuce and green leaf lettuce require four weeks to reach marketable size. The cultivation of four growth stages of these lettuce varieties allows one fourth of the crop to be harvested weekly. In three years of continuous operation of UVI's commercial-scale system, 148 crops of lettuce were harvested, which demonstrates the system's sustainability. Leafy green vegetables, herbs and other crops with short production periods are well suited for continuous, staggered production systems.

A batch cropping system is more appropriate for crops that are grown seasonally or have long growing periods (>3 months) such as tomatoes and cucumbers. Various intercropping systems can be used in conjunction with batch cropping. For example, if lettuce is intercropped with tomatoes and cucumbers, one crop of lettuce can be harvested before tomato plant canopy development limits light availability (Resh, 1995).

19.13 Pest and Disease Control

A number of plant pest and disease problems have been encountered in aquaponic systems. Pests observed on tomatoes include spider mite (Landesman, 1977; Nair et al. 1985), russet mite (Rakocy, unpublished data), hornworm (Lewis et al. 1978; Sutton and Lewis, 1982; Nair et al. 1985), western locust (Sutton and Lewis, 1982), fall armyworm, pinworm, aphid, and leaf minor (Nair et al. 1985). Diseases observed on

tomatoes include blight (Lewis et al. 1980) and bacteria wilt (McMurtry et al. 1990). In UVI's systems, lettuce is affected by fall armyworm, corn earworm, and two species of pathogenic root fungus (*Pythium dissotocum* and *P. myriotylum*). The root diseases that plague conventional hydroponics may be a threat to aquaponics. Four viral, two bacterial and 20 fungal pathogens have been associated with root diseases in hydroponically grown vegetables (Stanghellini and Rasmussen, 1994). Most of the destructive root diseases in hydroponics have been attributed to the fungal genera *Pythium, Phytophthora, Plasmopara, Olpidium* and *Fusarium*.

Pesticides should not be used to control insects on aquaponic plant crops. Even pesticides that are registered would pose a threat to the fish and would not be permitted in a fish culture system. Similarly, most therapeutants for treating fish parasites and diseases should not be used either. Vegetables may absorb and concentrate them. Even the common practice of adding salt to treat fish diseases or reduce nitrite toxicity would be deadly to vegetables. Non-chemical methods of plant pest and disease control are required such as biological control (resistant cultivars, predators, antagonistic organisms, pathogens), physical barriers, traps, treatment of the nutrient solution (filtration, UV sterilization), manipulation of the physical environment and other specialized cultural practices. Opportunities for biological control methods are greater in enclosed greenhouse environments than exterior installations. McMurtry (1989) used *Encarsia formosa* and *Chrysopa carnea* to control greenhouse white fly (*Trialeurodes vaporariorum*) and *Hippodamia convergens* to control potato aphid (*Macrosiphum euphorbiae*). In UVI's systems, caterpillars are effectively controlled by twice-a-week spraying with *Bacillus thuringiensis*, a bacterial pathogen that is specific to caterpillars. The fungal root pathogens that are encountered in summer dissipate in winter in response to lower water temperature and manipulation of suspended solids levels. An outbreak of *Pythium* coincided with a period during which the efficiency of suspended solids removal was dramatically increased.

Prohibition on the use of pesticides makes crop production in aquaponic systems more difficult. However, this restriction assures that crops from aquaponic systems will be raised in an environmentally sound manner, free of pesticide residues. A major advantage of aquaponic systems is that crops are less susceptible to attack from soil-borne diseases. It also appears that aquaponic systems may be more resistant to diseases that

affect standard hydroponics. This resistance may be due to the presence of some organic matter in the culture water which creates a stable, ecologically-balanced, growing environment with a wide diversity of microorganisms, some of which may be antagonistic to plant root pathogens.

19.14 Approaches to System Design

Several approaches can be used to design an aquaponic system. The simplest approach is to duplicate a standard system or scale a standard system down or up, keeping the components proportional. Changing aspects of a standard system is not recommended because changes often lead to unintended consequences. However, the design process often starts with a production goal for either fish or plants. In those cases there are some guidelines which can be followed.

Use a standard system that is already designed.

The easiest approach is to use a standard system design that has been tested and is in common use with a good track record. It is early in the development of aquaponics, but standard designs will emerge. The UVI system has been well documented and is being studied or used commercially in several locations, but there are other systems with potential. Standard designs will include specifications for layout, tank sizes, pipe sizes, pipe placement, pumping rates, aeration rates, infrastructure needs, etc. There will be operation manuals and projected production levels and budgets for various crops. Using a standard design will reduce risk.

Design for available space.

If a limited amount of space is available such as in an existing greenhouse, then that space will define the size of the aquaponic system. The easiest approach is to take a standard design and scale it down. If a scaled-down tank or pipe size falls between commercially available sizes, it is best to select the larger size. However, the water flow rate should equal the scaled-down rate for best results. The desired flow rate can be obtained by buying a higher capacity pump and installing a bypass line and valve, which circulates a portion of the flow back to the sump and allows the desired flow rate to go from the pump to the next stage of the system. If more space is available than the standard design requires, then the system could be scaled up within limitations or more than one scaled-down system could be installed.

Design for fish production

If the primary objective is to produce a certain amount of fish annually, the first step in the design process will be to determine the number of systems required, the number of rearing tanks required per system and the optimum rearing tank size. The number of harvests will have to be calculated based on the length of the culture period. Assume that the final density is 60 kg/m^3 (0.5 lbs/gallon) for an aerated system. Take the annual production per system and multiply it by the estimated feed conversion ratio (the kilograms of feed required to produce one kilogram of fish). Convert the pounds of annual feed consumption to grams (454 g/lb) and divide by 365 days to obtain the average daily feeding rate. Divide the average daily feeding rate by the desired feeding rate ratio, which ranges from 60 to 100 g/m^2/day for raft culture, to determine the required plant production area. For other systems such as NFT, the feeding rate ratio should be decreased in proportion to the water volume reduction of the system as discussed in the component ratio section. Use a ratio near the low end of the range for small plants such as Bibb lettuce and a ratio near the high end of the range for larger plants such as Chinese cabbage or romaine lettuce. The solids removal component, water pump, and blowers should be sized accordingly

Sample problem:

This example illustrates only the main calculations, which are simplified (e.g., mortality is not considered) for the sake of clarity. Assume that you have a market for 227 kg (500 lbs) of live tilapia per week in your city and that you want to raise lettuce with the tilapia because there is a good market for green leaf lettuce in your area. The key questions are: How many UVI aquaponic systems do you need to harvest 227 kg (500 lbs) of tilapia weekly. How large should the rearing tanks be? What is the appropriate number and size of hydroponic tanks? What would the weekly lettuce harvest be?

1) Each UVI system contains four fish rearing tanks (Fig. 9.3). Fish production is staggered so that one fish tank is harvested every 6 weeks. The total growing period per tank is 24 weeks. If 227 kg (500 lbs) of fish are required weekly, six production systems (24 fish rearing tanks) are needed.

2) Aquaponic systems are designed to achieve a final density of 60 kg/m^3 (0.5 lb/gallon). Therefore the water volume of the rearing tanks is 3.79 m^3 (1,000 gallons).

3) In 52 weeks, there will be 8.7 harvests (52/6 = 8.7) per system. Annual production for the system therefore is 2.0 mt (4,350 lbs) i.e. 227 kg per harvest x 8.7 harvests.

4) The usual feed conversion ratio is 1.7. Therefore annual feed input to the system is 3,360 kg (7,395 lbs) i.e. 2.0 mt x 1.7 = 3,360 kg.

5) The average daily feed input is 9.22 kg (20.3 lbs) i.e. 3,360 kg per year/365 days= 9.22 kg.

6) The average daily feed input converted to grams is 9,216 g i.e. 9.22 kg x 1000 g/kg = 9,216 g).

7) The optimum feeding rate ratio for raft aquaponics ranges from 60 – 100 g/m^2/day. Select 80 g/m^2/day as the design ratio. Therefore, the required lettuce growing area is 115.2 m^2 (9216 g/day divided by 80 g/m^2/day = 115.2 m^2).

8) The growing area in square feet is 1,240 (115.2 m^2 x 10.76 ft^2/m^2 = 1,240 ft^2).

9) Select a hydroponic tank width of 1.22 m (4 ft). Therefore, the total length of the hydroponic tanks is 94.5 m (310 ft) i.e. 115.2 m^2 /1.22 m = 94.5 m).

10) Select four hydroponic tanks. They are 23.6 m (77.5 ft) long (94.5/4 = 23.6 m). They are rounded up to 24.4 m (80 ft) in length, which is a practical length for a standard greenhouse and allows the use of ten 2.36 m (8 ft) sheets of polystyrene per hydroponic tank.

11) Green leaf lettuce produces good results with plant spacing of 48 plants per sheet (16/m^2). The plants require a 4-week growth period. With staggered production, one hydroponic tank is harvested weekly. Each hydroponic tank with 10 polystyrene sheets produces 480 plants. With six aquaponic production systems 2,880 plants are harvested weekly.

In summary, weekly production of 227 kg (500 lbs) of tilapia results in the production of 2,880 green leaf lettuce plants (120 cases). Six aquaponic systems each with four 3.78 m^3 (1,000-gallon) rearing tanks (water volume) are required. Each system will have four raft hydroponic tanks that are 24.4 m long and 1.22 m wide (80 ft long by 4 ft wide).

DESIGN FOR PLANT PRODUCTION

If the primary objective is to produce a certain quantity of plant crops annually, the first step in the design process will be to determine the area required for plant production. The area needed will be based on plant spacing, length of the production cycle, number of crops per year or growing season, and the estimated yield per unit area and per crop cycle. Select the desired feeding rate ratio and multiple by the total area to obtain the average daily feeding rate that is required. Multiply the average daily feeding rate by 365 days to determine annual feed consumption. Estimate the feed conversion ratio (FCR) for the fish species that will be cultured. Convert FCR to feed conversion efficiency. For example, if FCR is 1.7:1, then the feed conversion efficiency is 1 divided by 1.7 or 0.59. Multiply the annual feed consumption by the feed conversion efficiency to determine net annual fish yield. Estimate the average fish weight at harvest and subtract the anticipated average fingerling weight at stocking. Divide this number into the net annual yield to determine the total number of fish produced annually. Multiply the total number of fish produced annually by the estimated harvest weight to determine total annual fish production. Divide total annual fish production by the number of production cycles per year. Take this number and divide by 60 kg/m^3 (0.5 lb/gallon) to determine the total volume that must be devoted to fish production. The required water volume can be partitioned among multiple systems and multiple tanks per system with the goal of creating a practical system size and tank array. Divide the desired individual fish weight at harvest by 60 kg/m^3 (0.5 lb/gallon) to determine the volume of water required per fish. Divide the volume of water required per fish into the water volume of the rearing tank to determine the fish stocking rate. Increase this number by 5 to 10% to allow for expected mortality during the production cycle. The solids removal component, water pump, and blowers should be sized accordingly.

Sample problem:

Assume that there is a market for 1,000 Bibb lettuce plants weekly in your city. These plants will be sold individually in clear plastic clamshell

containers. A portion of the root mass will be left intact to extend self life. Bibb lettuce transplants are cultured in a UVI raft system for 3 weeks at a density of 29.3 plants/m^2. Assume that tilapia will be grown in this system. The key questions are: How large should the plant growing area be? What will be the annual production of tilapia? How large should the fish rearing tanks be?

1) Bibb lettuce production will be staggered so that 1,000 plants can be harvested weekly. Therefore, with a 3-week growing period, the system must accommodate the culture of 3,000 plants.

2) At a density of 29.3 plants/m^2, the total plant growing area will be 102.3 m^2 (3000 plants divided 29.3/m^2 = 102.3 m^2). This area is equal to 1,100 square feet; i.e. 102.3 m^2 x 10.76 ft^2/m^2 = 1,100 ft^2.

3) Select a hydroponic tank width of 2.44 m (8 ft). Therefore, the total hydroponic tank length will be 41.9 m (137.5 ft); i.e.102.3 m^2 /2.44 m = 41.9 m.

4) Two raft hydroponic tanks are required for the UVI system. Therefore the minimum length of each hydroponic tank will be 20.95 m (68.75 ft) i.e. 41.9 m /2 = 20.95 m). Since polystyrene sheets come in 2.44 (8 ft) lengths, the total number of sheets per hydroponic tank will be 8.59 sheets (20.95 m divided by 2.44 m/sheet = 8.59 sheets). To avoid wasting material, round up to nine sheets. Therefore, the hydroponic tanks will be 21.94 m (72 ft) long; i.e.9 sheets x 2.44 m per sheet = 21.94 m).

5) The total plant growing area will then be 107 m^2 (1,152 ft^2); i.e. 21.94 m x 2.44 m per tank x 2 tanks = 107 m^2.

6) At planting density of 29.3 plants/m^2, a total of 3,135 plants will be cultured in the system. The extra plants will provide a safety margin against mortality and plants that do not meet marketing standards.

7) Assume that a feeding rate ration of 60 g/m^2/day provides sufficient nutrients for good plant growth. Therefore, daily feed input to the system will be 6,420 g (14.1 lbs); i.e. 60 g/m^2/day x 107 m^2 = 6,420 g.

8) Annual feed input to the system will be 2,340 kg (5,146 lbs) i.e. 6.42 kg/day x 365 days = 2,340 kg)

9) Assume the feeding conversion ratio is 1.7. Therefore, the feed conversion efficiency is 0.59; i.e.1 kg of gain divided by 1.7 kg of feed = 0.59.

10) The total annual fish production gain will be 1,380 kg (3,036 lbs); i.e. 2,340 kg x 0.59 feed conversion efficiency = 1,380 kg.

11) Assume that the desired harvest weight of the fish will be 500 g (1.1 lbs) and that 50 g (0.11 lb) fingerlings will be stocked. Therefore, individual fish will gain 450 g (500 g harvest weight – 50 g stocking weight = 450 g). The weight gain per fish will be approximately 454 g (1 lb).

12) The total number of fish harvested will be 3,036; i.e. 1,380 kg of total gain divided by 0.454 kg of gain per fish = 3,036 fish.

13) Total annual production will be 1,518 kg (3,340 lbs) (3,036 fish x 0.50 kg/fish = 1,518 kg) when the initial stocking weight is considered.

14) If there are four fish rearing tanks and one tank is harvested every 6 weeks, there will be 8.7 harvests per year (52 weeks divided by 6 weeks = 8.7).

15) Each harvest will be 175 kg (384 lbs); i.e. 1,518 kg per year divided by 8.7 harvests per year = 175 kg/harvest).

16) Final harvest density should not exceed 60 kg/m^3 (0.5 lb/gallon). Therefore the water volume of each rearing tank should be 2.92 m^3 (768 gallons). The tank should be larger to provide a 2.4 cm (6 in) freeboard (space between the top edge of the tank and the water levels. A standard tank size of 3.8 m^3 (1,000 gallons) is recommended.

17) Assuming a mortality of 10% during the growth cycle, the tanks should be stocked with 385 juveniles each time ((175 kg / 0.5 kg/fish) x 1.1))

In summary, two hydroponic tanks each 21.94 m (72 ft) long by 2.44 m (8 ft) wide will be required to produce 1,000 Bibb lettuce plants per week. Four fish rearing tanks with a water volume of 3.8 m^3 (1,000 gallons) per tank will be required. Approximately 175 kg (384 lbs); of tilapia will be harvested every 6 weeks, and annual tilapia production will be 1,380 kg (3,036 lbs).

19.15 Economics

The economics of aquaponic systems depend on specific site conditions and markets. It would be inaccurate to make sweeping generalizations because material costs, construction costs, operating costs and market prices vary by location. For example, an outdoor tropical system would be less expensive to construct and operate than a controlled-environment greenhouse system in a cold temperate climate. Nevertheless the economic potential of aquaponic systems looks promising based on studies with the UVI system in the Virgin Islands and in Alberta, Canada.

The UVI system is capable of producing approximately 5,000 kg (11,000 lbs) of tilapia and 1,400 cases of lettuce or 5,000 kg (11,000 lbs) basil annually based on studies in the Virgin Islands. Enterprise budgets for tilapia production combined with either lettuce or basil have been developed. The U.S. Virgin Islands represent a small niche market with very high prices for fresh tilapia, lettuce, and basil as more than 95% of vegetables supplies and nearly 80% of fish supplies are imported. The budgets were prepared to show revenues, costs, and profits from six production units. A commercial enterprise consisting of six production units is recommended because one fish-rearing tank (out of 24) could be harvested weekly, thereby providing a continuous supply of fish for market development.

The enterprise budget for tilapia and lettuce show that the annual return to risk and management (profit) for six production units is US$185,248. The sale prices for fish $1.14/kg ($2.50/lb) and lettuce $20.00/case have been established through many years of market research at UVI. Most of the lettuce consumed in the Virgin Islands is imported from California. It is transported by truck across the United States to East Coast ports and then shipped by ocean freighters to Caribbean islands. Local production capitalizes on the high price that transportation adds to imported lettuce. Local production surpasses the quality of imported lettuce due to its freshness. Although this enterprise budget is unique to the U.S. Virgin Islands, it indicates that aquaponic systems can be profitable in certain niche markets.

The enterprise budget for tilapia and basil shows that the annual returns to risk and management for six production units are US$693,726. Aquaponic systems are very productive in producing culinary herbs such as basil. A conservative sales price for fresh basil with stems in the U.S. Virgin Islands is $4.55/kg ($10.00/lb). However, this enterprise budget is not realistic in terms of market demand. The population (108,000 people) of the U.S. Virgin Islands cannot absorb 30,000 kg (66,000 lbs) of fresh basil annually, although there are opportunities for provisioning ships and export to neighboring islands. A more realistic approach for a six-unit operation is to devote a portion of the growing area to basil to meet local demand while growing other crops in the remainder of the system.

The breakeven price for the aquaponic production of tilapia in the Virgin Islands is $0.67/kg ($1.47/lb) compared to a sales price of $1.14/kg ($2.50/lb). The breakeven prices are $6.15/case for lettuce (sales price = S20.00/case) and $0.34/kg ($0.75/lb) for basil (sales price $4.55/kg). The breakeven prices for tilapia and lettuce do not compare favorably to commodity prices. However, the cost of construction materials, electricity, water, labor, and land are very high in the U.S. Virgin Islands. Breakeven prices for tilapia and lettuce could be considerably lower in other locations. The breakeven price for basil compares favorably to commodity prices because fresh basil has a short shelf life and cannot be shipped great distances.

A UVI aquaponics system in an environmentally-controlled greenhouse at the Crops Diversification Center South in Alberta, Canada was evaluated for the production of tilapia and a number of plant crops. The crops were cultured for one production cycle and their yields were extrapolated to annual production levels. Based on prices at the Calgary wholesale market, annual gross revenue was determined for each crop per unit area and per system with a plant growing area of 250 m^2 (2,690 ft^2) (Table 19.8).

Annual production levels based on extrapolated data from short production cycles are subject to variation. Similarly, wholesale prices will fluctuate during the year based on supply and demand. Nevertheless the data indicates that culinary herbs in general can obtain a gross income more than 20 times greater than that of fruiting crops such as tomatoes and cucumbers. It appears that just one production unit could provide a livelihood for a small producer. However, this data does not show capital, operating, and marketing costs, which will be considerable. Furthermore, the quantity of herbs produced could flood the market and depress prices. Competition from current market suppliers will also lead to price reductions.

Table 19.8 Preliminary Production and Economic Data from the UVI Aquaponic System at the Crop Diversification Center South, Alberta, Canada.[1]
(Data courtesy of Dr. Nick Savidov)

Crop	Annual Production		Wholesale Price		Total Value	
	lb/ft^2	tons/2690 ft^2	Unit	$	$/ft^2	$/2690 ft^2
Tomatoes	6.0	8.1	15 lb	17.28	6.90	18,542
Cucumbers	12.4	16.7	2.2 lb	1.58	8.90	23,946
Egg Plant	2.3	3.1	11 lb	25.78	5.33	14,362
Genovese Basil	6.2	8.2	3 oz	5.59	186.64	502,044
Lemon Basil	2.7	3.6	3 oz	6.31	90.79	244,222
Osmin Basil	1.4	1.9	3 oz	7.03	53.23	143,2[illegible]8
Cilantro	3.8	5.1	3 oz	7.74	158.35	425,959
Parsley	4.7	6.3	3 oz	8.46	213.81	575,162
Portulaca	3.5	4.7	3 oz	9.17	174.20	468,618

[1]Ecomonic data based on Calgary wholesale market prices for the week ending July 4, 2003.

19.16 Prospects For The Future

Aquaponics is still in its infancy is becoming very popular in recent years and is being practiced mainly at the hobby and backyard levels. It is estimated that there are 1,500 aquaponic systems in the U.S. and many times this level in Australia. However, the number of commercial operations is still relatively small in the U.S. Hydroponic growers generally do not consider aquaculture as a nutrient source for their operations. Aquaculturists, on the other hand, frequently mention the possibility of incorporating hydroponics into their closed recirculating systems to mitigate waste discharge and earn extra income. Data from successful, large-scale trials is needed to attract investor capital and spur commercial development.

Although the design principles of aquaponic systems and the choice of hydroponic components and fish and plant combinations may seem challenging, aquaponic systems are quite simple to operate when fish are stocked at a rate that provides a good feeding rate ratio for plant production. Aquaponic systems are easier to operate than hydroponic systems or recirculating fish production systems because less monitoring is required and there is generally a wider safety margin for ensuring good water quality. Small aquaponic systems can provide an excellent hobby. Systems can be as small as an aquarium with a tray of plants covering the aquarium top. Large commercial operations comprised of many production units and occupying several acres are certainly possible if markets can absorb the output. The educational potential of aquaponic systems is already being realized in hundreds of schools where students learn a wide range of subjects that are demonstrated through the construction and operation of aquaponic systems. Regardless of scale or purpose, the culture of fish and plants through aquaponics is a gratifying endeavor that provides food.

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Table A-1a Mass Conversion Factors

Multiply Number of	By	To Obtain
grams	15.43	grains
grams	0.03527396	ounces (avoirdupois)
grams	0.03215075	ounces (troy)
grams	0.002205	pounds
kilograms	**2.205**	**pounds**
kilograms	9.84E-04	tons (long)
kilograms	1.10E-03	tons (short)
ounces	437.5	grains
ounces	28.319523	grams
ounces	0.0625	pounds
ounces	0.9115	ounces (troy)
pounds	7,000	grains
pounds	**453.5924**	**grams**
pounds	0.4536	kilograms
pound	16	ounces
pounds (troy)	373.24177	grams
tons (long)	1,016	kilograms
tons (long)	**2240**	**pounds**
tons (long)	1.12	tons (short)
tons (metric)	**2,205**	**pounds**
tons (short)	907.1848	kilograms
tons (short)	**2,000**	**pounds**
tons (short)	0.89287	tons (long)
tons (short)	0.90718	tons (metric)
To Obtain	Divide By	Starting With

[a]Units on the right column may be obtained by starting with units on the left column, **multiply by the given number.** To go the other way, from right to left, divide by the given value. Example: kilograms = 0.4536 * pounds and pounds = kilograms / 0.4536

Table A-1b Length Conversion Factors

Multiply Number of	By	To Obtain
centimeters	**0.3937**	**inches**
centimeters	0.03281	feet (ft)
centimeters	0.01094	yards
fathoms	6	feet
fathoms	1.828	meters
feet	304.8	millimeters
feet	**30.48**	**centimeters**
feet	0.3048	meters
inches	25.4	millimeters
inches	**2.54**	**centimeters**
inches	0.083333	feet
inches	0.0254	meters
inches	0.02777	yards
kilometers	3280.8	feet
kilometers	1,094	yards
kilometers	0.62137	miles
meters	**39.37**	**inches**
meters	**3.281**	**feet**
meters	1.0937	yards
meters	5.40E-04	miles(nautical)
meters	6.21E-04	miles(statute)
micron (μm)	3.937E-05	inches
micron (μm)	1.00E-06	meters
mil	0.001	inches
miles(nautical)	6080.27	feet
miles(nautical)	1.853	kilometers
miles(statute)	5280	feet
miles(statute)	1.609	kilometers
miles(statute)	0.8684	miles(nautical)
millimeters	0.03937	inches
millimeters	3.28E-03	feet
yards	91.44	centimeters
yards	0.9144	meters
To Obtain	Divide By	Starting With

[a]Units on the right column may be obtained by starting with units on the left column, multiply by the given number. To go the other way, from right to left, divide by the given value. Example: feet = 30.48 * centimeters and centimeters = feet / 30.48

Table A-1c Area Conversion

Multiply Number of	By	To Obtain
acres	0.4047	hectares
acres	**43,560**	**sq. ft.**
acres	4,047	sq. meters
acres	0.001562	sq. miles
hectares	2.471	acres
hectares	107,639	sq. ft.
square centimeters	1.076E-03	sq. ft.
square centimeters	0.155	sq. in.
square centimeters	0.0001	sq. in.
square feet	929	sq. cm
square feet	**0.0929**	**sq. meters**
square feet	0.1111	sq. yards
square inches	**6.452**	**sq. cm**
square inches	645.2	sq. millimeters
square kilometers	247.1	acres
square kilometers	1.076E+07	sq. ft.
square kilometers	1.00E+06	sq. meters
square kilometers	**0.3861**	**sq. miles**
square meters	**10.76**	**sq. ft.**
square meters	1,550	sq. in.
square meters	3.86E-07	sq. miles
square meters	1.196	sq. yards
square miles	**640**	**acres**
square miles	2.79E+07	sq. ft.
square miles	**2.59**	**sq. km**
square millimeters	1.55E-3	sq. in.
square yards	8,361	sq. cm
To Obtain	Divide By	Starting With

[a]Units on the right column may be obtained by starting with units on the left column, multiply by the given number. To go the other way, from right to left, divide by the given value. Example: square feet = 10.76 * square meters and square meters = square feet / 10.76

Th8able A-1d Volume Conversion Factors

Multiply Number of	By	To Obtain
acre-feet	43,560	cubic feet
acre-feet	325,850	gallons
cubic centimeters	3.53E-05	cubic feet
cubic centimeters	0.0610	cubic inches
cubic feet	28,320	cubic cm
cubic feet	1,728	cubic inches
cubic feet	0.02832	cubic meters
cubic feet	**7.48052**	**gallons (US liq.)**
cubic feet	**28.317**	**liters**
cubic inches	16.39	cubic centimeters
cubic inches	0.0005787	cubic feet
cubic inches	1.633E-05	cubic meters
cubic inches	0.004329	gallons
cubic meters	**35.315**	**cubic feet**
cubic meters	61,023	cubic inches
cubic meters	1.308	cubic yards
cubic meters	**264.17**	**gallons (US liq.)**
gallons	3,785,412	cubic cm
gallons	**0.1337**	**cubic feet**
gallons	231	cubic inches
gallons	0.004951	cubic yards
gallons	**3.785**	**liters**
gallons (liq. British imp.)	1.20095	gallons (US liq.)
gallons (US liq.)	0.83267	gallons (liq. British imp.)
liter	**0.03531**	**cubic feet**
liter	61.023	cubic inches
liter	0.2642	gallons(US liq.)
quarts (US)	0.9463	liters
tablespoons (US)	14.79	milliliters
teaspoons (US)	4.93	milliliters
To Obtain	Divide By	Starting With

[a]Units on the right column may be obtained by starting with units on the left column, multiply by the given number. To go the other way, from right to left, divide by the given value. Example: liters = 3.785 * gallons and gallons = liters / 3.785

Table A-1e Flow Rate Conversion Factors

Multiply Number of	By	To Obtain
cubic feet/min	472	ml/sec
cubic feet/min	0.125	gallons/sec
cubic feet/min	28.31	liters/min
cubic feet/min	**1.699**	**m^3/hr**
cubic feet/min	0.1247	gallons/sec
cubic feet/min	0.472	liters/sec
cubic feet/min	62.43	lbs of H_2O/min
cubic feet/min	**4.72E-04**	**m^3/s**
cubic feet/sec	0.6463	million gallons/day
cubic feet/sec	**448.831**	**gallons/min**
cubic feet/sec	28.317	liters/sec
cubic feet/sec	0.02832	m^3/s
cubic meters/min	0.01667	m^3/s
cubic meters/day	264.17	gallons/day
cubic meters/day	0.0002642	million gallons/day
cubic meters/sec	**35.3147**	**cu.ft./sec**
cubic meters/sec	22.82	million gallons/day
cubic meters/sec	15,850	gallons/min
gallons/day	4.38E-08	m^3/s
gallons/hr	63.08	ml/min
gallons/hr	0.1337	cu.ft./hr
gallons/min	0.002228	cu.ft./sec
gallons/min	**0.06308**	**liters/sec**
gallons/min	**8.0208**	**cu.ft./hr**
gallons/min	0.227	m^3/hr
gallons/min	**3.785**	**liters/min**
gallons/min	6.31E-05	m^3/s
liter/hr	2.78E-07	m^3/s
liters/min	1.667E-05	m^3/s
liters/min	0.2642	gallons/min
liters/min	4.403E-03	gallons/sec
liters/sec	15.84	gallons/min
liters/sec	0.0228	million gallons/day
million gallons/day	1.54723	cu.ft./sec
million gallons/day	694.4	gallons/min
To Obtain	Divide By	Starting With

[a]Units on the right column may be obtained by starting with units on the left column, multiply by the given number. To go the other way, from right to left, divide by the given value. Example: gpm = 448.831 * cfs and cfs = gpm / 448.831

Table A-1f Velocity Conversion Factors

Multiply Number of	By	To Obtain
cm/sec	1.969	ft/min
cm/sec	0.03281	ft/sec
cm/sec	0.036	km/hr
cm/sec	0.02237	miles/hr
cm/sec	0.0003728	miles/min
feet/min	0.508	cm/sec
feet/min	0.01667	feet/sec
feet/min	0.01136	miles/hr
feet/sec	**30.48**	**cm/sec**
feet/sec	1.097	km/hr
feet/sec	18.29	meters/min
feet/sec	0.6818	meters/hr
feet/sec	0.01136	miles/min
km/hr	27.78	cm/sec
km/hr	54.68	ft/min
km/hr	0.6214	miles/hr
meters/min	1.667	cm/sec
meters/min	3.281	ft/min
meters/min	0.5468	ft/sec
meters/min	0.03728	miles/hr
meters/sec	196.8	feet/min
meters/sec	**3.281**	**ft/sec**
meters/sec	3.6	km/hr
meters/sec	2.237	miles/hr
meters/sec	0.03728	miles/min
miles/hr	44.7	cm/sec
miles/hr	88	feet/min
miles/hr	1.467	feet/sec
miles/hr	**1.609**	**km/hr**
miles/hr	0.2682	km/min
miles/hr	26.82	meters/min
miles/hr	0.01667	miles/min
miles/min	2682	cm/sec
miles/min	88	feet/sec
miles/min	1.609	km/min
To Obtain	Divide By	Starting With

[a]Units on the right column may be obtained by starting with units on the left column, multiply by the given number. To go the other way, from right to left, divide by the given value. Example: cm/sec = 30.48 * ft/sec and ft/sec = cm/sec / 30.48

Table A-1g Pressure Conversion Factors

Multiply Number of	By	To Obtain
atmospheres	760	mm of mercury
atmospheres	**33.957**	**ft of water**
atmospheres	29.921	in of mercury
atmospheres	1.0333	kg/sq. cm
atmospheres	**14.696**	**pounds/sq.inch**
bars	0.9869	atmospheres
bars	14.5036	pound/sq. inch
bars	1,000,000	dynes/sq. cm
bars	10,197	kg/sq. meter
mm of mercury	0.001316	atmospheres
mm of mercury	0.04468	feet of water
mm of mercury	0.01934	pounds/sq. inch
feet of water	0.0295	atmospheres
feet of water	0.8811	inches of mercury
feet of water	304.8	kg/sq. meter
feet of water	**62.3205**	**pounds/sq. ft**
inches of mercury	0.03342	atmospheres
inches of mercury	1.1349	feet of water
inches of mercury	0.4912	pounds/sq. inch
inches of water	0.002454	atmospheres
inches of water	0.07343	inches of mercury
inches of water	5.193	pounds/sq. ft
inches of water	**0.03613**	**pounds/sq. inch**
kg/sq. cm	32.81	feet of water
kg/sq. cm	28.96	inches of mercury
kg/sq. cm	2.048	pounds/sq. ft
kg/sq. cm	14.22	pounds/sq. in.
pounds/sq. foot	4.73E-04	atmospheres
pounds/sq. foot	0.01602	feet of water
pounds/sq. foot	4.882	kg/sq. meter
pounds/sq. foot	6.94E-03	pounds/sq. inch
pounds/sq. inch	0.06804	atmospheres
pounds/sq. inch	2.3068	**feet of water**
pounds/sq. inch	2.036	inches of mercury
pounds/sq. inch	27.7276	inches of water
To Obtain	Divide By	Starting With

[a]Units on the right column may be obtained by starting with units on the left column, multiply by the given number. To go the other way, from right to left, divide by the given value. Example: feet of water = 2.307 * psi and psi = feet of water / 2.307

Table A-2 Fish Health Conversion Factors

1 ppm (mg/L)	=	0.38 grams per 100 gallons of water
		3.8 milligrams per gallon of water
		0.0283 grams per cubic foot of water
		0.38 milliliters per 100 gallons of water
		2.72 pounds per acre-foot of water
		1 milligrams per liter of water (mg/L)
		1 grams per cubic meter of water (g/m^3)
		0.001 milliliters per liter of water (mL/L)
1 acre-foot	=	43,560 cubic feet
1 acre-foot	=	325,850 gallons
1 acre foot of water	=	2,718,144 pounds
1 cubic foot of water	=	7.48 gallons
1 cubic foot of water	=	62.4 pounds
1 cubic foot of water	=	28.3 liters
1 cubic foot of water	=	28.3 kilograms
1 cubic meter of water	=	1,000 liters
1 cubic meter of water	=	35.32 cubic feet
1 cubic meter of water	=	2,205 pounds
1 gallon of water	=	8.34 pounds
1 gram	=	0.0353 ounces
1 kilogram	=	2.2 pounds
1 pound	=	454 grams
1 gallon	=	3.785 liters
1 gallon of water	=	3,785 grams
1 liter	=	0.26 gallons
1 liter	=	1,000 cubic centimeters
1 liter	=	1,000 milliliters
1 liter of water	=	1,000 grams
1 ounce	=	28.35 grams
1 gallon	=	128 fluid ounces
1 fluid ounce	=	28.4 grams
1 inch	=	2.54 centimeters
1 foot	=	30.48 centimeters
1 cubic centimeter of water	=	1 gram
1 cubic centimeter of water	=	1 milliliter
1 hectare	=	10,000 square meters
1 hectare	=	2.47 acre
1 acre	=	0.405 hectare
1 acre	=	43,560 square feet
Percent Solution		
For 1 percent solution add:		
38 grams per gallon		1.3 ounces per gallon
10 grams per liter		38 cc per gallon
10 cc per liter		
Temperature Conversion		
Centigrade to Fahrenheit	=	(°C X 9/5) + 32
Fahrenheit to Centigrade	=	(°F - 32) X 5/9

Table A-3 Miscellaneous List of Conversion Factors: Inch:Pound (IP) to System International (SI)

Power		
1 Btu/s	1.0551	kW
1 Btu/min	17.59	W
1 Btu/hr	0.22931	W
1 ft-lb/s	1.3558	W
1 ft-lb/min	0.0260	W
1 ft-lb/hr	3.767E-04	W
1 horsepower (mechanical)	0.7457	kW
1 horsepower (boiler)	9.810	kW
1 hp-h	2.6845	MJ
1 ton (refrigeration, 12,000 Btu/hr)	3.517	kW
1 J/s	1.00	W
Energy		
1 Btu	1.0551	kJ
1 ft-lb	1.356	J
1 horsepower-hr	2.685	MJ
1 kWh	3.600	MJ
1 calorie (gram)	4.187	J
1 therm	105.5	MJ
1 langley	4.186	J/cm^2
1 langley	41.86	kJ/m^2
1 quad (US)	1.055E+12	MJ
Mass per unit time, flow rate		
1 lb/s	0.4536	kg/s
1 lb/min	7.560E-03	kg/s
1 lb/hr	1.260E-04	kg/s
1 lb/hr	5.515E-04	tonne/hr
1 ton/hr	0.2520	kg/s
1 ton/hr	1.103	tonne/hr
Force, weight		
1 cwt	444.8	N
1 kip (= 1000 lb)	4,448	N
1 lb	4.448	N
1 ounce	0.2780	N
1 stone	62.27	N
1 ton	9,964	N

Fan efficiency		
1 cfm/watt	0.4719	m^3-s/kW
1 cfm/watt	4.719E-04	m^3-s/W
Thermal conductivity		
1 Btu/hr-ft-F	1.731	W/m-K
1 Btu-in/hr-ft^2-F	0.1442	W/m-K
Thermal conductance		
1 Btu/hr-ft^2-F	5.678	W/m^2-K
Thermal resistance		
1 hr-ft^2-F/Btu	0.1761	m^2-K/W
Thermal flux, solar radiation		
1 Btu/hr-ft^2	3.169	W/m^2
1 langley.min	698	W/m^2
1 langley	41,860	J/m^2
1 langley	41.86	kJ/m^2
1 Btu/ft^2	11,400	J/m^2
1 Btu/ft^2	11.40	kJ/m^2
1 Btu/ft^2	0.0114	MJ/m^2
Light level, illumination		
1 foot-candle	10.76	Lux
Photosynthetically Active Radiation (PAR)		
1 Btu/hr-ft^2 (solar)	6.565	mmol/m^2-s (solar)
1 Btu/ft^2 (solar)	0.02363	mol/m^2 (solar)
Heat value (as of fuels)		
1 Btu/ft^3	37.26	kJ/m^3
1 Btu/gal	0.2785	kJ/L
1 Btu/lb	2.326	kJ/kg
Heat capacity		
1 Btu/lb-F	4,187	J/kg-K
1 Btu/lb-F	4.187	kJ/kg-K

Table A-4 (Table 2.1) Physical Properties of Water

Temp. C	Density kg/m^3	Kinematic Viscosity (m^2/s)·E-06	Vapor Pressure mm Hg
0	999.84	1.79	4.8
1	999.90	1.73	5.1
2	999.94	1.68	5.4
3	999.97	1.62	5.8
4	1000.00	1.57	6.2
5	999.97	1.52	6.6
6	999.94	1.48	7.0
7	999.90	1.43	7.5
8	999.85	1.39	8.0
9	999.78	1.35	8.5
10	999.70	1.31	9.1
11	999.61	1.27	9.7
12	999.50	1.23	10.3
13	999.38	1.20	11.0
14	999.25	1.17	11.7
15	999.10	1.13	12.5
16	998.94	1.10	13.4
17	998.78	1.07	14.3
18	998.60	1.05	15.2
19	998.41	1.02	16.2
20	998.21	0.99	17.3
21	997.99	0.97	18.4
22	997.77	0.95	19.7
23	997.54	0.93	21.0
24	997.30	0.90	22.4
25	997.05	0.88	23.9
26	996.79	0.87	25.5
27	996.52	0.85	27.2
28	996.24	0.83	29.0
29	995.95	0.81	30.9
30	995.65	0.80	33.0
31	995.34	0.78	35.2
32	995.03	0.77	37.5
33	994.71	0.75	40.0
34	994.38	0.74	42.7
35	994.04	0.72	45.5
36	993.69	0.71	48.5
37	993.33	0.70	51.8
38	992.97	0.68	55.2
39	992.60	0.67	58.9
40	992.22	0.66	62.8

Table A-5 (Table 2.5) Percentage of Free Ammonia (as NH_3) in Freshwater at Varying pH and Water Temperature, (Spotte, 1979)

pH	10°C (50°F)	15°C (59°F)	20°C (68°F)	25°C (77°F)	30°C (86°F)
6.5	0.06	0.09	0.13	0.18	0.25
6.6	0.07	0.11	0.16	0.23	0.32
6.7	0.09	0.14	0.20	0.28	0.40
6.8	0.12	0.17	0.25	0.36	0.50
6.9	0.15	0.22	0.31	0.45	0.63
7.0	0.19	0.27	0.39	0.56	0.80
7.1	0.23	0.34	0.50	0.71	1.00
7.2	0.29	0.43	0.63	0.89	1.26
7.3	0.37	0.54	0.79	1.12	1.58
7.4	0.46	0.68	0.99	1.40	1.98
7.5	0.59	0.86	1.24	1.76	2.48
7.6	0.74	1.07	1.56	2.21	3.10
7.7	0.92	1.35	1.96	2.76	3.87
7.8	1.16	1.69	2.45	3.45	4.82
7.9	1.46	2.12	3.06	4.31	6.00
8.0	1.83	2.65	3.83	5.37	7.43
8.1	2.29	3.32	4.77	6.66	9.18
8.2	2.86	4.14	5.94	8.25	11.3
8.3	3.58	5.16	7.36	10.0	
8.4	4.46	6.41	9.09	12.3	16.8
8.6	6.88		13.6		24.2
8.8	10.5		20.0		33.6
9.0	15.6		28.4		44.5
9.2	22.7		38.5		56.0
9.4	31.8		49.8		66.8
9.6	42.5		61.2		76.2

Table A-6 Dissolved Oxygen (mg O_2 per Liter, ppm) at Saturation in Water of Various Temperature and Salinity (Colt, 1984 [1])

Temp	Salinity, parts per thousand								
(°C)	0	5	10	15	20	25	30	35	40
0	14.602	14.112	13.638	13.180	12.737	12.309	11.896	11.497	11.111
1	14.198	13.725	13.268	12.825	12.398	11.984	11.585	11.198	10.825
2	13.813	13.356	12.914	12.487	12.073	11.674	11.287	10.913	10.552
3	13.445	13.004	12.576	12.163	11.763	11.376	11.003	10.641	10.291
4	13.094	12.667	12.253	11.853	11.467	11.092	10.730	10.380	10.042
5	12.757	12.344	11.944	11.557	11.183	10.820	10.470	10.131	9.802
6	12.436	12.036	11.648	11.274	10.911	10.560	10.220	9.892	9.573
7	12.127	11.740	11.365	11.002	10.651	10.311	9.981	9.662	9.354
8	11.832	11.457	11.093	10.742	10.401	10.071	9.752	9.443	9.143
9	11.549	11.185	10.833	10.492	10.162	9.842	9.532	9.232	8.941
10	11.277	10.925	10.583	10.252	9.932	9.621	9.321	9.029	8.747
11	11.016	10.674	10.343	10.022	9.711	9.410	9.118	8.835	8.561
12	10.766	10.434	10.113	9.801	9.499	9.207	8.923	8.648	8.381
13	10.525	10.203	9.891	9.589	9.295	9.011	8.735	8.468	8.209
14	10.294	9.981	9.678	9.384	9.099	8.823	8.555	8.295	8.043
15	10.072	9.768	9.473	9.188	8.911	8.642	8.381	8.129	7.883
16	9.858	9.562	9.276	8.998	8.729	8.468	8.214	7.968	7.730
17	9.651	9.364	9.086	8.816	8.554	8.300	8.053	7.814	7.581
18	9.453	9.174	8.903	8.640	8.385	8.138	7.898	7.664	7.438
19	9.201	8.990	8.726	8.471	2.222	7.982	7.748	7.521	7.300
20	9.077	8.812	8.556	8.307	8.065	7.831	7.603	7.382	7.167
21	8.898	8.641	8.392	8.149	7.914	7.685	7.463	7.248	7.038
22	8.726	8.476	8.233	7.997	7.767	7.545	7.328	7.118	6.914
23	8.560	8.316	8.080	7.849	7.626	7.409	7.198	6.993	6.794
24	8.400	8.162	7.931	7.707	7.489	7.277	7.072	6.872	6.677
25	8.244	8.013	7.788	7.569	7.357	7.150	6.950	6.754	6.565
26	8.094	7.868	7.649	7.436	7.229	7.027	6.831	6.641	6.456
27	7.949	7.729	7.515	7.307	7.105	6.908	6.717	6.531	6.350
28	7.808	7.593	7.385	7.182	6.984	6.792	6.606	6.424	6.248
29	7.671	7.462	7.259	7.060	6.868	6.680	6.498	6.321	6.148
30	7.539	7.335	7.136	6.943	6.755	6.572	6.394	6.221	6.052
31	7.411	7.212	7.018	6.829	6.645	6.466	6.293	6.123	5.959
32	7.287	7.092	6.903	6.718	6.539	6.364	6.194	6.029	5.868
33	7.166	6.976	6.791	6.611	6.435	6.265	6.099	5.937	5.779
34	7.049	6.863	6.682	6.506	6.335	6.168	6.006	5.848	5.694
35	6.935	6.753	6.577	6.405	6.237	6.074	5.915	5.761	5.610
36	6.824	6.647	6.474	6.306	6.142	5.983	5.828	5.676	5.529
37	6.716	6.543	6.374	6.210	6.050	5.894	5.742	5.594	5.450
38	6.612	6.442	6.277	6.117	5.960	5.807	5.659	5.514	5.373
39	6.509	6.344	6.183	6.025	5.872	5.723	5.577	5.436	5.297
40	6.410	6.248	6.091	5.937	5.787	5.641	5.498	5.360	5.224

[1] Colt, J., 1984. Computation of dissolved gas concentrations in water as functions of temperature, salinity, and pressure. American Fisheries Society Special Publication, 14, Bethesda, Maryland.

Table A-7 Hardness Conversion to Other Units of Measure (gr is the abbreviation for grain and 1 g = 15.43 gr)

Units of Measure	mg/L $CaCO_3$	English gr/gal (Imperial) $CaCO_3$	American gr/gal (US) $CaCO_3$	French parts/ 100,000 $CaCO_3$	German parts/ 100,000 CaO	meq/L	g/L CaO	lb/cu ft $CaCO_3$
mg/L$CaCO_3$	1.0	0.07	0.058	0.1	0.056	0.02	5.64×10^{-4}	6.23×10^{-5}
English gr/gal (Imperial) $CaCO_3$	14.3	1.0	0.83	1.43	0.83	0.286	8.0×10^{3}	8.91×10^{-4}
American gr/gal (US) $CaCO_3$	17.1	1.2	1.0	1.72	0.96	0.343	9.66×10^{-3}	1.07×10^{-3}
French parts/100,000 $CaCO_3$	10.0	0.7	0.58	1.0	0.56	0.2	5.6×10^{-3}	6.23×10^{-4}
German parts/100,000 CaO	17.9	1.25	1.04	1.79	1.0	0.358	1.0×10^{-2}	1.12×10^{-3}
meq/L	50	3.5	2.9	5.0	2.8	1.0	2.8×10^{-2}	3.11×10^{-3}
g/L CaO	1,790	125	104.2	179	100	35.8	1.0	0.112
lb/cu ft $CaCO_3$	16,100	1.123	935	1,610	900	321	9.0	1.0

Table A-8 (Table 3.7) Standard U.S. Atmospheric Pressure at Different Altitudes

Altitude		Atmospheric Pressure				
M	ft	mm Hg	kPa abs	psia	m H_2O	ft H_2O
-200	-656	778	103.7	15.0	10.6	34.8
0	0	760	101.3	14.7	10.3	33.8
200	656	742	98.9	14.3	10.1	33.1
400	1,312	725	96.6	14.0	9.9	32.5
600	1,968	707	94.3	13.7	9.6	31.5
800	2,625	690	92.0	13.3	9.4	30.8
1,000	3,281	674	89.8	13.0	9.2	30.2
4,000	13,123	462	61.6	8.9	6.3	20.7

Table A-9 (Table 2.6) Alkalinity Supplement Properties (Bisogni and Timmons, 1991)

Chemical Formula	Common Name(s)	Equivalent Wt. (gm/eq.)	Solubility	Rate of solubilization
NaOH	sodium hydroxide	40	high	rapid
Na_2CO_3	sodium carbonate soda ash	53	high	rapid
$NaHCO_3$	sodium bicarbonate baking soda	83	high	rapid
$CaCO_3$	calcium carbonate calcite	50	moderate	moderate
CaO	slaked lime	28	high	moderate
$Ca(OH)_2$	calcium hydroxide hydrated lime	37	high	moderate
$CaMg(CO_3)_2$	dolomite	46	moderate	slow
$MgCO_3$	magnesium carbonate magnesite	42	moderate	slow
$Mg(OH)_2$	magnesium hydroxide brucite	29	moderate	slow

Note: Na compounds are highly soluble in water while Mg compounds have poor solubility. Ca compounds are intermediate. Mg compounds tend to dissolve very slowly, so may have application in situations requiring a long-term application. Na compounds may prove to be the most expensive.

Based on 100% pure compound. To calculate for impurities, divide the tabulated value by the pure fraction ({100%-impurities %}/100 gives pure fraction) to get the true value.

Table A-10 (Table 8.13) Opening Sizes of U.S. Sieve Series Designation Number (Perry and Chilton, 1973)

Sieve Designation Number†	Size of Opening, (mm)	Sieve Designation Number†	Size of Opening (mm)
4	4.76	35	0.500
5	4.00	40	0.420
6	3.36	45	0.354
7	2.83	50	0.297
8	2.38	60	0.250
10	2.00	70	0.210
12	1.68	80	0.177
14	1.41	100	0.149
16	1.19	120	0.125
18	1.00	140	0.105
20	0.841	170	0.088
25	0.707	200	0.074
30	0.595	230	0.063

† Number of meshes per inch.

Table A-11 (Table 10.1) Dry Air Components

Species	% volume	% mass	Molecular Wt.
Nitrogen	78.084	75.600	28.0
Oxygen	20.946	23.200	32.0
CO_2	0.032	0.048	44.0
Argon	0.934	1.300	39.9
Air	100.000	100.000	29.0

Table A 12 (Table 10.2) Solubility of Four Major Gases in Water

Gas Species	Solubility in Air* (mg/L)	Solubility of pure gas* (mg/L)
Oxygen	10.08	48.14
Nitrogen	16.36	20.95
Argon	0.62	65.94
Carbon dioxide	0.69	1992.00

*At 15 Deg. C.

Table A-13 Maximum and Minimum Monthly Average Outside Temperatures for Selected USA Locations in Degrees °F (Kreider, J. F., & Kreith, F. ,1975, Solar Heating And Cooling: Engineering, Practical Design, And Economics, Washington, DC: Hemisphere Publishing)

Location	Max	Min	Location	Max	Min
Mobile, AL	82.6	53.0	Great Falls, MT	69.4	22.1
Phoenix, AZ	89.8	49.7	Omaha, NE	78.5	22.3
Little Rock, AR	81.9	40.6	Reno, NV	67.7	30.4
Los Angeles, CA	69.1	54.4	Concord, NH	69.6	21.2
Denver, CO	72.9	28.5	Atlantic City, NJ	75.1	34.7
Hartford, CT	73.4	26.0	Albuquerque, NM	78.5	35.0
Wilmington, DE	76.0	33.4	Buffalo, NY	69.8	24.1
Washington, DC	78.2	36.9	Raleigh, NC	77.9	41.6
Miami, FL	82.3	66.9	Bismark, ND	71.7	9.9
Atlanta, GA	78.9	44.7	Columbus, OH	74.8	29.9
Honolulu, HI	79.4	72.4	Oklahoma City, OK	82.8	37.0
Boise, ID	75.2	29.1	Portland, OR	67.2	38.4
Chicago, IL	75.6	26.0	Pittsburgh, PA	72.1	28.9
Indianapolis, IN	75.2	29.1	Providence, RI	72.1	29.2
Des Moines, IA	76.3	19.9	Columbia, SC	81.6	46.4
Wichita, KS	80.9	32.0	Sioux Falls, SD	74.3	15.2
Louisville, KY	77.6	35.0	Memphis, TN	81.3	41.5
New Orleans, LA	81.9	54.6	Dallas, TX	85.0	45.9
Portland, ME	68.1	21.8	Salt Lake City, UT	76.9	27.2
Baltimore, MD	76.8	34.8	Burlington, VT	69.0	16.2
Boston, MA	73.7	29.9	Richmond, VA	78.1	38.7
Detroit, MI	74.4	26.9	Seattle, WA	64.9	38.3
Minneapolis, MN	72.3	12.4	Charleston, WV	74.9	36.6
Jackson, MS	82.3	47.9	Milwaukee, WI	68.7	20.6
St. Louis, MO	78.1	31.9	Cheyenne, WY	70.0	25.4

Table A-14 Unit Area Thermal Resistance's (R-Values) of Typical Building Construction and Insulation Materials (ASHRAE, 1981, Handbook of Fundamentals. Atlanta, GA: American Society of Heating, Refrigerating, and Air-Conditioning Engineers)

Building Material	R-value °F-ft^2·hr/BTU (W/m^{2}K)
Insulation, per inch	
Fiberglass Batt	4.00
Cellulose	3.1-3.7
Mineral Wool	2.5-3.0
Sawdust	2.22
Expanded Polystyrene	5.00
Extruded Rubber	4.55
Polyisocyanurate	7.04
Foam-in-place Polyurethane	6.00
Wood Materials, per inch	
Softwoods (spruce, pine, fir)	1.25
Hardwoods	.91
Plywood	1.25
Cinder Block, total (per inch)	
4 inch thick	1.1(0.19)
8 inch thick	1.7 (0.30)
12 inch thick	1.9 (0.33)
Concrete, per inch	.08
Asphalt shingles, total (per inch)	0.4 (0.07)
Other Materals	
Gypsum board, ½"	.45
Lapped Wood Siding, ½" x 8"	.81
Metal Siding, hollow backed	.61
Air Space, ¾ to 4"	.90
Convection Coefficients for Wall Surfaces[1]	
Winter time 15 mph (24 kph) wind	0.17 (0.029)
Summer time 7.5 mph (12 kph) wind	0.25 (0.044)
Still air (less than 100 ft/min or 0.5 m/s)	1.00 (0.176)

[1]Convection coefficients are the inverse of these R values; to convert (°F-ft^2-hr/BTU) to m^{2}K/W divide by 5.678

Table A-15 Allowable Concentrations of Carbon Dioxide at Standard Temperature and Pressure to the Nearest 25 ppm (ASHRAE, 1991, Handbook - Applications. Atlanta, GA: American Society of Heating, Refrigerating, and Air-Conditioning Engineers)

Toxicity Standard	Allowable Concentration (ppm)
Immediately dangerous to life and health[a]	50,000
Acceptable ceiling concentration, not to be exceeded during an 8 hour shift[b]	30,000
Time-weighted average, not to be exceeded in any 8 hour shift of a 40 hour week[b]	5,000
Atmospheric concentration	350

[a]National Institute for Occupational Safety and Health standard
[b]Occupational Safety and Health Administration standard

Table A-16 Valve Basics and Selection Tips

Gate Valves

Gate values are designed to operate fully open or fully closed. Because they operate slowly, they prevent fluid hammer, which is detrimental to piping systems. There is very little pressure loss through a fully open gate valve.

Ball Valves

Ball valves are also designed to be operated fully open or fully closed with any liquid containing particles that could scratch the ball. Many people use them successfully for throttling clear water. Ball valves have low pressure drops, open and close quickly, are simple, and are usually trouble free. With the development of Teflon seals, ball valves have grown in popularity. One problem with ball valves is that opening and closing them quickly can cause fluid hammer.

Butterfly Valves

Butterfly valves, like ball valves, operate with a ¼ turn. They are generally used for handling large flows of gasses or liquids, and should not be used for throttling for extended periods of time. They are also very compact relative to flanged gate and ball valves.

Globe Valves

Globe valves advantages are that they close slowly to prevent fluid hammer, the flow can be throttled and they will not leak under low pressure when they are shut off. Flow and pressure control valves as well as hose bibs generally use the globe pattern. The disadvantage of this design is that the "Z" pattern of flow restricts flow more than gate, ball or butterfly valves.

Table A-17 Plastic Properties for Various Types of Pipes

PVC

Polyvinyl Chloride, Type 1, Grade 1. This pipe is strong, rigid and resistant to a variety of acids and bases. Some solvents and chlorinated hydrocarbons may damage the pipe. PVC is very common, easy to work with and readily available at commercial vendors. Maximum useable temperature is 140°F (60°C) and pressure ratings start at a minimum of 125 to 200 psi (8 ATM to 13 ATM) (check for specific ratings stamped on the pipe). PVC can be used with water, gas, and drainage systems, but **NOT** for hot water or pressurized gases.

ABS

Acrylonitrite Butadiene Styrene, Type 1. This pipe is strong and rigid and resistant to a variety of acids and bases. Some solvents and chlorinated hydrocarbons may damage the pipe. ABS is very common, easy to work with and readily available from commercial vendors. Maximum useable temperature is 160°F (71°C) at low pressure. It is most commonly used as a DWV pipe.

CPVC

Chlorinated polyvinyl chloride. Similar to PVC, but designed specifically for piping water at up to 180°F (82°C), although it can withstand 200°F for a limited time. Pressure rating is 100 psi (7 ATM). Note diameter and fittings are not interchangeable with PVC pipe and fitting.

PE

Polyethylene. A flexible pipe for pressurized water systems, not for hot water.

PB

Polybutylene. A flexible pipe for pressurized water systems, both hot and cold. ONLY compression and banded type joints can be used.

Table A-18 Size and Properties of PVC Pipe Schedule 40 and 80

Nominal Size (in)	Actual OD (in)	PVC Schedule 40 Inside Diameter (in)	PVC Schedule 40 Wall Thickness (in)	PVC Schedule 40 Weight (lbs/ft)	PVC Schedule 80 Inside Diameter (in)	PVC Schedule 80 Wall Thickness (in)	PVC Schedule 80 Weight (lbs/ft)
¼	0.540				0.302	0.119	0.10
½	0.840	0.622	0.109	0.16	0.546	0.147	0.21
¾	1.050	0.824	0.113	0.22	0.742	0.154	0.28
1	1.315	1.049	0.133	0.32	0.957	0.179	0.40
1¼	1.660	1.380	0.140	0.43	1.278	0.191	0.57
1½	1.900	1.610	0.145	0.52	1.500	0.200	0.69
2	2.375	2.067	0.154	0.70	1.939	0.218	0.95
2½	2.875	2.469	0.203	1.10	2.323	0.276	1.45
3	3.500	3.068	0.216	1.44	2.900	0.300	1.94
4	4.500	4.026	0.237	2.05	3.826	0.337	2.83
6	6.625	6.065	0.280	3.61	5.761	0.432	5.41
8	8.625	7.981	0.322	5.45	7.625	0.500	8.22
10	10.750	10.020	0.365	7.91	9.564	0.593	12.28
12	12.750	11.938	0.406	10.35	11.373	0.687	17.10

Table A-19 (Table 12.3) PVC Pipe Friction Chart for Pipe and Length Loss for Schedule 40

GPM	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)
		1/2 inch			3/4 inch			1 inch			1-1/4 inch			1-1/2 inch			2 inch			3 inch	
1	1.13	2.08	0.90	0.63	0.51	0.22															
2	2.26	4.18	1.80	1.26	1.02	0.44	0.77	0.55	0.24	0.44	0.14	0.06									
5	5.64	23.44	10.15	3.16	5.73	2.48	1.93	1.72	0.75	1.11	0.44	0.19	0.81	0.22	0.09						
7	7.90	43.06	15.64	4.43	10.52	4.56	2.72	3.17	1.37	1.55	0.81	0.35	1.13	0.39	0.17						
10				6.32	20.36	8.82	3.86	6.02	2.61	2.21	1.55	0.67	1.62	0.72	0.31	0.98	0.21	0.09			
15				9.48	42.46	18.39	5.79	12.77	5.53	3.31	3.28	1.42	2.42	1.53	0.66	1.46	0.45	0.19			
20							7.72	21.75	9.42	4.42	5.59	2.42	3.23	2.61	1.13	1.95	0.76	0.33	0.88	0.11	0.05
25										5.52	8.45	3.66	4.04	3.95	1.71	2.44	1.15	0.50	1.10	0.17	0.07
30		4 inch								6.63	11.85	5.13	4.85	5.53	2.39	2.93	1.62	0.70	1.33	0.23	0.10
35										7.73	15.76	6.82	5.66	7.36	3.19	3.41	2.15	0.93	1.55	0.31	0.13
40	1.02	0.11	0.05							8.84	20.18	8.74	6.47	9.43	4.08	3.90	2.75	1.19	1.77	0.40	0.17
45	1.15	0.13	0.06		5 inch								7.27	11.73	5.08	4.39	3.43	1.49	1.99	0.50	0.22
50	1.28	0.16	0.07										8.08	14.25	6.17	4.88	4.16	1.80	2.21	0.60	0.26
60	1.53	0.22	0.10	0.97	0.07	0.03										5.85	5.84	2.53	2.65	0.85	0.37
70	1.79	0.30	0.13	1.14	0.10	0.04		6 inch								6.83	7.76	3.36	3.09	1.13	0.49
75	1.92	0.34	0.15	1.22	0.11	0.05										7.32	8.82	3.82	3.31	1.28	0.55
80	2.05	0.38	0.16	1.30	0.13	0.06	0.90	0.05	0.02							7.80	9.94	4.30	3.53	1.44	0.62
90	2.30	0.47	0.20	1.46	0.16	0.07	1.01	0.06	0.03							8.78	12.37	5.36	3.98	1.80	1.78
100	2.56	0.58	0.25	1.62	0.19	0.08	1.12	0.08	0.04		8 inch					9.75	15.03	6.51	4.42	2.18	1.94
125	3.20	0.88	0.38	2.03	0.29	0.13	1.41	0.12	0.05										5.52	3.31	1.43
150	3.84	1.23	0.53	2.44	0.40	0.17	1.69	0.16	0.07	0.97	0.04	0.02							6.63	4.63	2.00
175	4.48	1.63	0.71	2.84	0.54	0.24	1.97	0.22	0.10	1.14	0.06	0.03		10 inch					7.73	6.16	2.67
200	5.11	2.08	0.90	3.25	0.69	0.30	2.25	0.28	0.12	1.30	0.07	0.03							8.83	7.88	3.41
250	6.40	3.15	1.36	4.06	1.05	0.45	2.81	0.43	0.19	1.63	0.11	0.05	1.03	0.04	0.02		12 inch				
300	7.67	4.41	1.91	4.87	1.46	0.63	3.37	0.60	0.26	1.94	0.15	0.07	1.23	0.05	0.02						
350	8.95	5.87	2.55	5.69	1.95	0.85	3.94	0.79	0.34	2.27	0.21	0.09	1.44	0.07	0.03	1.01	0.03	0.01			
400	10.23	7.52	3.26	6.50	2.49	1.08	4.49	1.01	0.44	2.59	0.27	0.12	1.64	0.09	0.04	1.16	0.04	0.02			
450				7.31	3.09	1.34	5.06	1.26	0.55	2.92	0.33	0.14	1.85	0.11	0.05	1.30	0.05	0.02			
500				8.12	3.76	1.63	5.62	1.53	0.66	3.24	0.40	0.17	2.05	0.13	0.06	1.45	0.06	0.03			
750							8.43	3.25	1.41	4.86	0.85	0.37	3.08	0.28	0.12	2.17	0.12	0.05			
1000							11.24	5.54	2.40	6.48	1.45	0.63	4.11	0.48	0.21	2.89	0.20	0.09			
1250										8.11	2.20	0.95	5.14	0.73	0.32	3.62	0.31	0.13			
1500										9.72	3.07	1.33	6.16	1.01	0.44	4.34	0.43	0.19			
2000													8.21	1.72	0.74	5.78	0.73	0.32			
2500													10.27	2.61	1.13	7.23	1.11	0.49			
3000																8.68	1.55	0.67			

(Shaded area recommended flow rates to minimize settlement of solids and avoid scouring of walls and junctions)

Table A-20 PVC Pipe Friction Chart for Pipe and Length Loss for Schedule 80

GPM	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)	Velocity (fps)	Head Loss (ft)	Head Loss (psi)
		1/2 inch			3/4 inch			1 inch			1-1/4 inch			1-1/2 inch			2 inch			3 inch	
1	1.48	4.02	1.74	0.74	0.86	0.37															
2	2.95	8.03	3.48	1.57	1.72	0.74	0.94	0.88	0.38	0.52	0.21	0.09	0.38	0.10	0.04						
5	7.39	45.23	19.59	3.92	9.67	4.19	2.34	2.75	1.19	1.30	0.66	0.29	0.94	0.30	0.13	0.56	0.10	0.04			
7	10.34	83.07	35.97	5.49	17.76	7.69	3.28	5.04	2.19	1.82	1.21	0.53	1.32	0.55	0.24	0.78	0.15	0.07			
10				7.84	33.84	14.65	4.68	9.61	4.16	2.60	2.30	1.00	1.88	1.04	1.45	1.12	0.29	0.13			
15				11.76	71.70	31.05	7.01	20.36	8.82	3.90	4.87	2.11	2.81	2.20	0.95	1.68	0.62	0.27			
20							9.35	34.68	15.02	5.20	8.30	3.59	3.75	3.75	1.62	2.23	1.06	0.46	1.00	0.15	0.07
25							11.69	25.43	22.70	6.50	12.55	5.43	4.69	5.67	2.46	2.79	1.60	0.69	1.25	0.22	0.10
30		4 inch								7.80	17.59	7.62	5.63	7.95	3.44	3.35	2.25	0.97	1.49	0.31	0.13
35										9.10	23.40	10.13	6.57	10.58	4.58	3.91	2.99	1.29	1.74	0.42	0.18
40	1.15	0.14	0.06							10.40	29.97	12.98	7.50	13.55	5.87	4.47	3.83	1.66	1.99	0.54	0.23
45	1.29	0.17	0.07		5 inch					11.70	37.27	16.14	8.44	16.85	7.30	5.03	4.76	2.07	2.24	0.67	0.29
50	1.43	0.21	0.09							13.00	45.30	19.61	9.38	20.48	8.87	5.58	5.79	2.51	2.49	0.81	0.35
60	1.72	0.30	0.13	1.08	0.10	0.04							11.26	28.70	12.43	6.70	8.12	3.52	2.99	1.14	0.49
70	2.01	0.39	0.17	1.26	0.13	0.06		6 inch								7.82	10.80	4.68	3.49	1.51	0.65
75	2.15	0.45	0.19	1.35	0.14	0.06										8.38	12.27	5.31	3.74	1.72	0.74
80	2.29	0.50	0.22	1.44	0.16	0.07	1.00	0.07	0.03							8.93	13.83	5.99	3.99	1.94	0.84
90	2.58	0.63	0.27	1.62	0.20	0.09	1.13	0.08	0.04							10.05	17.20	7.45	4.48	2.41	1.04
100	2.87	0.76	0.33	1.80	0.24	0.10	1.25	0.10	0.05		8 inch					11.17	20.90	9.05	4.98	2.93	1.27
125	3.59	1.16	0.50	2.25	0.37	0.16	1.57	0.16	0.07										6.23	4.43	1.92
150	4.30	1.61	0.70	2.70	0.52	0.23	1.88	0.22	0.10	1.07	0.05	0.02							7.47	6.20	2.68
175	5.02	2.15	0.93	3.15	0.69	0.30	1.20	0.29	0.12	1.25	0.08	0.03		10 inch					8.72	8.26	3.58
200	5.73	2.75	1.19	3.60	0.88	0.38	1.51	0.37	0.16	1.43	0.09	0.04							9.97	10.57	4.58
250	7.16	4.16	1.81	4.50	1.34	0.58	3.14	0.56	0.24	1.79	0.14	0.06	1.14	0.05	0.02		12 inch				
300	8.60	5.83	2.52	5.40	1.87	0.81	3.76	0.78	0.34	2.14	0.20	0.09	1.36	0.07	0.03						
350	10.03	7.76	3.36	6.30	2.49	1.08	4.39	1.04	0.45	2.50	0.27	0.12	1.59	0.09	0.04	1.12	0.04	0.02			
400	11.47	9.93	4.30	7.19	3.19	1.38	5.02	1.33	0.58	2.86	0.34	0.15	1.81	0.11	0.05	1.28	0.06	0.02			
450				8.09	3.97	1.72	5.64	1.65	0.71	3.21	0.42	0.18	2.04	0.14	0.06	1.44	0.06	0.03			
500				8.99	4.82	2.09	6.27	2.00	0.87	3.57	0.51	0.22	2.27	0.17	0.07	1.60	0.07	0.03			
750							9.40	4.25	1.84	5.36	1.08	0.47	3.40	0.36	0.16	2.40	0.15	0.07			
1000							12.54	7.23	3.13	7.14	1.84	0.80	4.54	0.61	0.26	3.20	0.26	0.11			
1250										8.93	2.78	1.20	5.67	0.92	0.40	4.01	0.40	0.17			
1500										10.71	3.89	1.68	6.8	1.29	0.56	4.81	0.55	0.24			
2000													9.07	2.19	0.93	6.41	0.94	0.41			
2500													11.34	3.33	1.44	8.01	1.42	0.62			
3000																9.61	1.99	0.86			

(Shaded area recommended flow rates to minimize settlement of solids and avoid scouring of walls and junctions)

Table A-21 Tank Volumes for Various Depths and Diameters

Diameter ft	Depth ft	Volume gallons	Volume m³	Diameter m	Depth m	Volume gallons	Volume m³
3	1	53	0.20	1	0.25	52	0.20
	2	106	0.40		0.5	104	0.39
	3	159	0.60		0.75	156	0.59
4	1	94	0.36	1.5	0.25	117	0.44
	2	188	0.71		0.5	233	0.88
	3	282	1.1		0.75	350	1.3
6	2	423	1.6	2	0.5	415	1.6
	3	635	2.4		0.75	622	2.4
	4	846	3.2		1	830	3.1
8	3	1128	4.3	2.5	0.5	648	2.5
	4	1504	5.7		0.75	973	3.7
	5	1880	7.1		1	1297	4.9
10	3	1763	6.7	3	0.75	1400	5.3
	4	2350	8.9		1	1867	7.1
	5	2938	11		1.25	2334	8.8
12	3	2538	10	4	0.75	2490	9.4
	4	3384	13		1	3320	13
	5	4230	16		1.25	4149	16
15	3	3966	15	5	0.75	3890	15
	4	5288	20		1	5187	20
	5	6609	25		1.25	6484	25
20	3	7050	27	6	0.75	5602	21
	4	9400	36		1	7469	28
	5	11750	44		1.25	9336	35
30	4	21150	80	10	4	82990	314
	6	31725	120		6	124484	471
	8	42300	160		8	165979	628
36	4	30456	115	12	4	119505	452
	6	45684	173		6	179258	679
	8	60912	231		8	239010	905
42	4	41454	157	13.5	4	151249	573
	6	62181	235		6	226873	859
	8	82908	314		8	302497	1145
48	4	54144	205	15	4	186727	707
	6	81216	307		6	280090	1060
	8	108288	410		8	373453	1414

To calculate the Volume of a Tank:

$$Volume(\,ft^3\,) = \frac{\pi D^2}{4} \cdot Depth$$

where Depth   Tank depth, ft

D   Tank diameter, ft

Conversions: 1 gal=7.481 ft^3; 1m^3=264.2 gal

Table A-22 Temperature Equivalents Between Celsius and Fahrenheit

C	F	C	F	C	F	C	F
0.0	32.0	**10.0**	50.0	**20.0**	68.0	**30.0**	86.0
0.6	33.0	10.6	51.0	20.6	69.0	30.6	87.0
1.0	33.8	**11.0**	51.8	**21.0**	69.8	**31.0**	87.8
1.1	34.0	11.1	52.0	21.1	70.0	31.1	88.0
1.7	35.0	11.7	53.0	21.7	71.0	31.7	89.0
2.0	35.6	**12.0**	53.6	**22.0**	71.6	**32.0**	89.6
2.2	36.0	12.2	54.0	22.2	72.0	32.2	90.0
2.8	37.0	12.8	55.0	22.8	73.0	32.8	91.0
3.0	37.4	**13.0**	55.4	**23.0**	73.4	**33.0**	91.4
3.3	38.0	13.3	56.0	23.3	74.0	33.3	92.0
3.9	39.0	13.9	57.0	23.9	75.0	33.9	93.0
4.0	39.2	**14.0**	57.2	**24.0**	75.2	**34.0**	93.2
4.4	40.0	14.4	58.0	24.4	76.0	34.4	94.0
5.0	41.0	**15.0**	59.0	**25.0**	77.0	**35.0**	95.0
5.6	42.0	15.6	60.0	25.6	78.0	35.6	96.0
6.0	42.8	**16.0**	60.8	**26.0**	78.8	**36.0**	96.8
6.1	43.0	16.1	61.0	26.1	79.0	36.1	97.0
6.7	44.0	16.7	62.0	26.7	80.0	36.7	98.0
7.0	44.6	**17.0**	62.6	**27.0**	80.6	**37.0**	98.6
7.2	45.0	17.2	63.0	27.2	81.0	37.2	99.0
7.8	46.0	17.8	64.0	27.8	82.0	37.8	100.0
8.0	46.4	**18.0**	64.4	**28.0**	82.4		
8.3	47.0	18.3	65.0	28.3	83.0		
8.9	48.0	18.9	66.0	28.9	84.0		
9.0	48.2	**19.0**	66.2	**29.0**	84.2		
9.4	49.0	19.4	67.0	29.4	85.0		

TABLE A-23 ELECTRICAL MEASUREMENTS

Voltage (volt) = Current (amp) * Resistance (ohm)
Power (watt) = Voltage (volt) * Current (amp)
Power (watt) = Current^2 (amp^2) * Resistance (ohm)

Single-phase AC motors:

$$horsepower(output) = \frac{volts * amps * Eff\% * PF}{746}$$

$$kilowatts = \frac{volts * amps * PF}{1{,}000}$$

Three-phase AC motors:

$$horsepower(output) = \frac{1.73 * volts * amps * Eff\% * PF}{746}$$

$$kilowatts = \frac{1.73 * volts * amps * PF}{1{,}000}$$

Water Pumps

$$horsepower(output) = \frac{GPM * Head\ in\ feet}{3{,}960 * Eff\ of\ pump}$$

Fans & Blowers

$$horsepower(output) = \frac{CFM * \text{Pressure}(psi)}{33{,}000 * Efficiency}$$

PF = Power Factor
EFF% = efficiency (0.5 to 0.85 for pumps)

Approximations: 110v motor draws 10 amps per HP
220v single-phase motor draws 5 amps per HP
220v three-phase motor draws 2.5 amps per HP
440v three-phase motor draws 1.25 amps per HP

Table A-24 Maximum length of wire in feet for 2% maximum voltage drop. If voltage drop is greater than 2%, efficiency of the equipment in the circuit is severely decreased and the life of the equipment will be decreased.

		Max Wire length in feet @ 120 volts, 1 phase, 2% max voltage drop						
Amps	Volt-Amps	#14	#12	#10	#8	#6	#4	#2
1	120	450	700	1100	1800	2800	4500	7000
5	600	90	140	225	360	575	910	1400
10	1200	45	70	115	180	285	455	705
15	1800	30	47	75	120	190	305	485
20	2400		36	57	90	140	230	365
25	3000			45	72	115	180	290
30	3600			38	60	95	150	240
40	4800				45	72	115	175
50	6000					57	90	145
60							76	120
70							65	105
80								90

		Max Wire length in feet @ 240 volts, 1 phase, 2% max voltage drop						
Amps	Volt-Amps	#14	#12	#10	#8	#6	#4	#2
1	240	900	1400	2200	3600	5600	9000	
5	1200	180	285	455	720	1020	1750	2800
10	2400	90	140	225	360	525	910	1400
15	3600	60	95	150	240	350	605	965
20	4800		70	110	180	265	455	725
25	6000			90	144	210	365	580
30	7200			75	120	175	300	485
40	9600				90	130	230	360
50	12000					105	180	290
60	14400						150	240
70	16800						130	205
80	19200							180

Table A-25 Current carrying capacity of hard-usage flexible cords (Type S, ST, SO, STO, SJ, SJT, SJO, SJTO)

Size Wire	Single-phase	Three-phase
18	10	7
16	13	10
14	18	15
12	25	20
10	30	25
8	40	35
6	55	45
4	70	60

Table A-26 Full load currents in amperes for single phase alternating-current motors.

HP	115 V	230V
¼	5.8	2.9
⅓	7.2	3.6
½	9.8	4.9
¾	13.8	6.9
1	16	8
1 ½	20	10
2	24	12
3	34	17
5	56	28
7 ½	80	40
10	100	50

Table A-27 Full load currents in amperes for three phase squirrel cage and wound rotor motors.

HP	Full load amps	Min. Wire Size
1	3.6	14
1.5	5.2	14
2	6.8	14
3	9.6	14
5	15.2	12
7.5	22	8
10	28	8
15	42	6

Table A-28 Recommended pipe sizes for standard air (0.075 lbs/ft3) for three pipe air velocities (Note the dynamic pressure for the three air velocities is 0.06, 0.25, and 1.00 inch WG for 1000, 2000, and 4000 fpm, respectively).

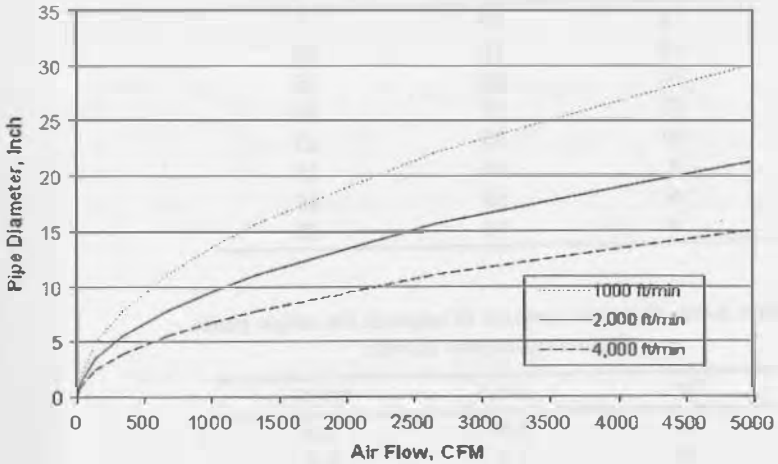


Table A-29 Area Under the Normal Distribution Curve

Z	0.00	0.01	0.02	0.03	0.04	0.05	0.06	0.07	0.08	0.09
0	.5000	.5040	.5080	.5120	.5160	.5199	.5239	.5279	.5319	.5359
0.1	.5398	.5438	.5437	.5517	.5557	.5596	.5636	.5675	.5714	.5753
0.2	.5793	.5832	.5871	.5910	.5948	.5987	.6026	.6064	.6103	.6141
0.3	.6179	.6217	.6255	.6293	.6331	.6368	.6406	.6443	.6480	.6517
0.4	.6554	.6591	.6628	.6664	.6700	.6736	.6772	.6808	.6844	.6879
0.5	.6915	.6950	.6985	.7019	.7054	.7088	.7123	.7157	.719	.7224
0.6	.7257	.7291	.7324	.7357	.7389	.7422	.7454	.7486	.7517	.7549
0.7	.7580	.7611	.7642	.7673	.7704	.7734	.7764	.7794	.7823	.7852
0.8	.7881	.7910	.7939	.7967	.7995	.8023	.8051	.8079	.8106	.8133
0.9	.8159	.8186	.8212	.8238	.8264	.8289	.8315	.8340	.8365	.8389
1.0	.8413	.8438	.8461	.8485	.8508	.8531	.8554	.8577	.8599	.8621
1.1	.8643	.8665	.8686	.8708	.8729	.8749	.8770	.8790	.8810	.8830
1.2	.8849	.8869	.8888	.8907	.8925	.8944	.8962	.8980	.8997	.9015
1.3	.9032	.9049	.9066	.9082	.9099	.9115	.9131	.9147	.9162	.9177
1.4	.9192	.9207	.9222	.9236	.9251	.9265	.9279	.9292	.9306	.9319
1.5	.9332	.9345	.9357	.9370	.9382	.9394	.9406	.9418	.9429	.9441
1.6	.9452	.9463	.9474	.9484	.9495	.9505	.9515	.9525	.9535	.9545
1.7	.9554	.9564	.9573	.9582	.9591	.9599	.9608	.9616	.9625	.9633
1.8	.9641	.9649	.9658	.9664	.9671	.9678	.9686	.9693	.9699	.9706
1.9	.9713	.9719	.9726	.9732	.9738	.9744	.9750	.9756	.9761	.9767
2.0	.9773	.9778	.9783	.9788	.9793	.9798	.9803	.9808	.9812	.9817
2.1	.9821	.9826	.9830	.9834	.9838	.9842	.9846	.9850	.9854	.9857
2.2	.9861	.9864	.9868	.9871	.9875	.9878	.9881	.9884	.9887	.9890
2.3	.9893	.9896	.9898	.9901	.9904	.9906	.9909	.9911	.9913	.9916
2.4	.9918	.9920	.9922	.9925	.9927	.9929	.9931	.9932	.9934	.9936
2.5	.9938	.9940	.9941	.9943	.9945	.9946	.9948	.9949	.9951	.9952
2.6	.9953	.9955	.9956	.9957	.9959	.9960	.9961	.9962	.9963	.9964
2.7	.9965	.9966	.9967	.9968	.9969	.9970	.0971	.9972	.9973	.9974
2.8	.9974	.9975	.9976	.9977	.9977	.9978	.9979	.9979	.9980	.9981
2.9	.9981	.9982	.9983	.9983	.9983	.9984	.9985	.9985	.9986	.9986
3.0	.99865	.99869	.99874	.99878	.99882	.99886	.99889	.99893	.99896	.99900
3.1	.99903	.99906	.99910	.99913	.99915	.99918	.99921	.99924	.99926	.99929
3.2	.99931	.99934	.99936	.99938	.99940	.99942	.99944	.99946	.99948	.99950
3.3	.99952	.99953	.99955	.99957	.99958	.99960	.99961	.99962	.99964	.99965
3.4	.99966	.99967	.99969	.99970	.99971	.99972	.99973	.99974	.99975	.99976
3.5	.99977	.99978	.99978	.99979	.99980	.99981	.99981	.99982	.99983	.99983

Z	0.00	0.01	0.02	0.03	0.04	0.05	0.06	0.07	0.08	0.09
-3.5	.00023	.00022	.00022	.00021	.00020	.00019	.00019	.00018	.00017	.00017
-3.4	.00034	.00033	.00031	.00030	.00029	.00028	.00027	.00026	.00025	.00024
-3.3	.00048	.00047	.00045	.00043	.00042	.00040	.00039	.00038	.00036	.00035
-3.2	.00069	.00066	.00064	.00062	.00060	.00058	.00056	.00054	.00052	.00050
-3.1	.00097	.00094	.00090	.00087	.00085	.00082	.00079	.00076	.00074	.00071
-3.0	.00135	.00131	.00126	.00122	.00118	.00114	.00111	.00107	.00104	.00010
-2.9	.0019	.0018	.0017	.0017	.0016	.0016	.0015	.0015	.0014	.0014
-2.8	.0026	.0025	.0024	.0023	.0023	.0022	.0021	.0021	.0020	.0019
-2.7	.0035	.0034	.0033	.0032	.0031	.0030	.0029	.0028	.0027	.0026
-2.6	.0047	.0045	.0044	.0043	.0041	.0040	.0039	.0038	.0037	.0036
-2.5	.0062	.0060	.0059	.0057	.0055	.0054	.0052	.0051	.0049	.0048
-2.4	.0082	.0080	.0078	.0075	.0073	.0071	.0069	.0068	.0066	.0064
-2.3	.0107	.0104	.0102	.0099	.0096	.0094	.0091	.0089	.0087	.0084
-2.2	.0139	.0136	.0132	.0129	.0125	.0122	.0119	.0116	.0113	.0110
-2.1	.0179	.0174	.0170	.0166	.0162	.0158	.0154	.0150	.0146	.0143
-2.0	.0228	.0222	.0217	.0212	.0207	.0202	.0197	.0192	.0188	.0183
-1.9	.0287	.0281	.0274	.0268	.0262	.0256	.0250	.0244	0.239	.0233
-1.8	.0359	.0351	.0344	.0336	.0329	.0322	.0314	.0307	.0301	.0294
-1.7	.0446	.0436	.0427	.0418	.0409	.0401	.0392	.0384	.0375	.0367
-1.6	.0548	.0537	.0526	.0516	.0505	.0495	.0485	.0475	.0465	.0455
-1.5	.0668	.0655	.0643	.0630	.0618	.0606	.0594	.0582	.0571	.0559
-1.4	.0808	.0793	.0778	.0764	.0749	.0735	.0721	.0708	.0694	.0681
-1.3	.0968	.0951	.0934	.0918	.0901	.0885	.0869	.0853	.0838	.0823
-1.2	.1151	.1131	.1112	.1093	.1075	.1057	.1038	.1020	.1003	.0985
-1.1	.1357	.1335	.1314	.1292	.1271	.1251	.1230	.1210	.1190	.1170
-1.0	.1587	.1562	.1539	.1515	.1492	.1469	.1446	.1423	.1401	.1379
-0.9	.1841	.1814	.1788	.1762	.1736	.1711	.1685	.1660	.1635	.1611
-0.8	.2119	.2090	.2061	.2033	.2005	.1977	.1949	.1922	.1894	.1867
-0.7	.2420	.2389	.2358	.2327	.2297	.2266	.2236	.2207	.2177	.2148
-0.6	.2743	.2709	.2676	.2643	.2611	.2578	.2546	.2514	.2483	.2451
-0.5	.3085	.3050	.3015	.2981	.2946	.2912	.2877	.2843	.2810	.2776
-0.4	.3446	.3409	.3372	.3336	.3300	.3264	.3228	.3192	.3156	.3121
-0.3	.3821	.3783	.3745	.3707	.3669	.3632	.3594	.3557	.3520	.3483
-0.2	.4207	.4168	.4129	.4090	.4052	.4013	.3974	.3936	.3897	.3859
-0.1	.4602	.4562	.4522	.4483	.4443	.4404	.4364	.4325	.4286	.4247
-0	.5000	.4960	.4920	.4880	.4840	.4801	.4761	.4721	.4681	.4641

Table A-30 Percentage Points of the Students t-Distribution

Percentage Points of Student's t-Distribution (value is area in one-tail of distribution curve)						
Degrees of Freedom	0.20	0.10	0.05	0.025	0.01	0.005
1	1.376	3.078	6.314	12.706	31.821	63.656
2	1.061	1.886	2.920	4.303	6.965	9.925
3	0.978	1.638	2.353	3.182	4.541	5.841
4	0.941	1.533	2.132	2.776	3.747	4.604
5	0.920	1.476	2.015	2.571	3.365	4.032
6	0.906	1.440	1.943	2.447	3.143	3.707
7	0.896	1.415	1.895	2.365	2.998	3.499
8	0.889	1.397	1.860	2.306	2.896	3.355
9	0.883	1.383	1.833	2.262	2.821	3.250
10	0.879	1.372	1.812	2.228	2.764	3.169
11	0.876	1.363	1.796	2.201	2.718	3.106
12	0.873	1.356	1.782	2.179	2.681	3.055
13	0.870	1.350	1.771	2.160	2.650	3.012
14	0.868	1.345	1.761	2.145	2.624	2.977
15	0.866	1.341	1.753	2.131	2.602	2.947
16	0.865	1.337	1.746	2.120	2.583	2.921
17	0.863	1.333	1.740	2.110	2.567	2.898
18	0.862	1.330	1.734	2.101	2.552	2.878
19	0.861	1.328	1.729	2.093	2.539	2.861
20	0.860	1.325	1.725	2.086	2.528	2.845
21	0.859	1.323	1.721	2.080	2.518	2.831
22	0.858	1.321	1.717	2.074	2.508	2.819
23	0.858	1.319	1.714	2.069	2.500	2.807
24	0.857	1.318	1.711	2.064	2.492	2.797
25	0.856	1.316	1.708	2.060	2.485	2.787
26	0.856	1.315	1.706	2.056	2.479	2.779
27	0.855	1.314	1.703	2.052	2.473	2.771
28	0.855	1.313	1.701	2.048	2.467	2.763
29	0.854	1.311	1.699	2.045	2.462	2.756
30	0.854	1.310	1.697	2.042	2.457	2.750
40	0.851	1.303	1.684	2.021	2.423	2.704
50	0.849	1.299	1.676	2.009	2.403	2.678
60	0.848	1.296	1.671	2.000	2.390	2.660
80	0.846	1.292	1.664	1.990	2.374	2.639
100	0.845	1.290	1.660	1.984	2.364	2.626
150	0.844	1.287	1.655	1.976	2.351	2.609
Infinity	0.842	1.282	1.645	1.960	2.326	2.576

DETERMINATION OF STATISTICAL DIFFERENCES: t-TEST FOR UNEQUAL VARIANCES

Because of natural biological variability, measurements of biological responses of replicated lots fish fed the same identical feed and subjected to identical fish cultural practices seldom give identical values. Therefore when testing new feeds or different cultural practices, it is important to compute the probability of any differences observed due to changes in feed or cultural operation to see such changes are simply a result of natural biological variability or whether they represent a real and statistically significant effect of instituting a change. One very useful statistic is the *t* test for two means having unequal variances. Most likely it will be necessary to obtain at least three replicated values for each mean in order to detect meaningful statistical differences; more is better. For instance one may make independent measurements during feeding of each of two different feeds to three separate lots of fish to obtain data or measurements of digestibility, growth rate, feed conversion, etc.

The *t*-test for unequal variances (also called Welch's *t* test) is an excellent statistical test to compare the difference between two means ($\mu_1 - \mu_2$) having unequal variances (s_1^2 and s_2^2). Seldom do means have truly equal variances. This test is better than the regular Student's *t*-test and the Mann-Whitney U test when variances are unequal (Ruxton 2006). All that is needed is the mean (μ), variance (s^2) and number of observations (n) for each of two means to be compared. For our purposes we will represent individual observations by the symbol y. For a more detailed discussion of this test see Ruxton (2006).

Calculation of *t* for the unequal variance *t*-test involves calculation of a *t* statistic that is compared with the appropriate value in standard *t* tables. The calculation of the unequal variance *t*-test (*t*) is given in Eq. 1:

$$t = \frac{(\mu_1 - \mu_2)}{\sqrt{\left(\frac{s_1^2}{n_1} + \frac{s_2^2}{n_2}\right)}} \qquad \text{(Eq. 1)}$$

Calculation of the degrees of freedom (v) for the unequal variance *t*-test, it is given by Eq. 2:

$$v = \frac{\left(\frac{1}{n_1} + \frac{u}{n_2}\right)^2}{\frac{1}{n_1^2(n_1-1)} + \frac{u^2}{n_2^2(n_2-1)}} \qquad \text{(Eq. 2)}$$

where u is computed in Eq. 3 as:

$$u = \frac{s_2^2}{s_1^2} \qquad \text{(Eq. 3)}$$

In general, v calculated from Equation 2 gives a non-integer value which is rounded down to the nearest whole number before consulting a standard *t* table.

Variance (s^2) for each mean is computed by Eq. 4:

$$s^2 = \frac{\Sigma y^2 - {(\Sigma y)^2}/{n}}{n-1} \qquad \text{(Eq. 4)}$$

$\sum y^2$ represents the uncorrected sum of squares (Uncor. SS) of each individual observation (y) constituting each mean (μ_1 and μ_2). $(\sum y)^2/n$ represents the correction term where n is the number of observations in each mean. $\sum y^2 - (\sum y)^2$ represents the corrected sum of squares (Corrected SS). The Corrected SS divided by n-1 equals variance (see below).

If *t* is approximately equal to zero, the indication is that $\mu_1 = \mu_2$. If *t* (based on $\mu_1 - \mu_2$) is much smaller than zero, the indication is that $\mu_1 < \mu_2$. If *t* is much larger than zero, the indication is that $\mu_1 > \mu_2$. By comparing the absolute value of the observed *t* from to the appropriate critical value given in a *t* distribution table (Appendix: Table A-30) one determines the probability of obtaining the difference observed between means by chance alone, i.e., the level of statistical significance.

The procedure of testing the hypothesis that the digestibilities of phosphorus in two diets (#1 and 2) are equal ($\mu_1 = \mu_2$) is illustrated by an example: Three measurements (n=3) of percentage digestibility (y) for diet #1 are 72.6, 71.9 and 73.3. For diet #2, the digestibilities were determined to 76.2, 75.2, and 77.4. The statistical question is to determine whether the mean digestibilities for the two diets are different.

THE PROCEDURE IS AS FOLLOWS:

1. Null Hypothesis: The two diet means are equal, that is, $\mu_1 = \mu_2$ (a 2-tailed t-test).
2. Alternative hypothesis: The alternative hypothesis is that (a) $\mu_1 < \mu_2$ or (b) $\mu_1 > \mu_2$.
3. Assumptions: The samples represent random and independent observations from normal populations with the unequal variances (s^2). The assumptions of randomness and independence of observations are very important. Independence of observations requires that there is no effect or influence of any observation upon the outcome of any of the other observations (replicates).
4. Level of significance: The 95% confidence level is chosen which means a difference of this magnitude occurs only 5% of the time simply by chance and normal biological variation when there is no significant or real difference.
5. Critical t value: The critical t value for a 2-tailed t test ($P<0.05$) with 3 degrees of freedom (computed as v using Equation 2) is the table value (3.182) found in the t-distribution table (Appendix: **Table A-30**). For a 2-tailed t-test at $P<0.05$, one uses a table value found in the column 0.025 (representing a total of 5% as the sum of 2.5% in each of two tails on the distribution).
6. Computation of t: The details of the computation of t (Equation 1) and v degrees of freedom (Equation 2) are given in table of calculations (below). The computed observed t-value is -4.78 with 3 degrees of freedom.

DEFINITIONS OF TERMS FOR THE CALCULATION OF t

Observation data values:	y
Number of observations:	n
Observed difference between sample means:	(μ_1 and μ_2)
Uncorrected sums of squares:	Uncor. SS = Σy^2
Correction term:	$(\Sigma y)^2/n$
Corrected sums of squares:	Cor. SS = $\Sigma y^2 - (\Sigma y)^2/n$
Variance:	Cor. SS/n-1

Table: Calculation of t-value for means with unequal variances

Item	Diet 1	Diet 2	Explanation
Observations (y)	72.9	76.4	Three independent random observations for each diet
	71.9	75.2	
	73.3	77.4	
Calculations:			
Σy	218.18	229.0	Sums
n	3	3	Number of observations
Means (μ)	72.700	76.333	(μ_1-μ_2=-3.633)
$(\Sigma y)^2$	47,567.61	52,441.00	Sums squared
$(\Sigma y)^2/n$	15,855.87	17,480.33	Correction term
Σy^2	15,856.91	17.482.76	Uncor. SS
SS	1.0400	2.4267	Cor. SS
s^2	0.52	1.2134	Variance

$t = 72.7 - 76.333/\sqrt{(0.52/3 + 1.2134/3)} = -4.78$ (from Eq. 1)

u = 1.2134/0.52 = 2.333 (from Eq. 3)

$v = (1/3 + 2.333/3)^2 / (1/3^2{*}2 + 2.333^2/3^2{*}2) = 3.4$ (from Eq. 2)

CONCLUSION:

In a 2-tailed t-test, the null hypothesis (H_o) would be that digestibility of phosphorus in diet 2 is not different from that for diet 1 (i.e., that $\mu_2 = \mu_1$), *verses* the alternative hypothesis (H_a) that the digestibility of phosphorus in diet 2 is different from that for diet 1 (i.e., that $\mu_2 > \mu_1$ or $\mu_2 < \mu_1$). The absolute value (4.78) of the observed t is greater than the critical table t (3.182) found in the 0.025 column of Table A-30 (Appendix) with 2.5% per each of two tails with 3 degrees of freedom. Because the observed t (4.78) is greater than the critical table t (3.182), we reject the null hypothesis and accept the alternative hypothesis and conclude using a 2-tailed test that μ_1 and μ_2 are significantly different at the 95% confidence level ($P < 0.05$).

In some cases there may be prior or logical reason to test a one directional change between means. For instance, one could modify the diet using a form of phosphorus in hopes a achieving a superior digestibility. In such a case, it is logical to ask a one-directional question. Thus a 1-tailed statistical *t*-test could be made such that the null hypothesis (H_o) could be that digestibility of phosphorus in diet 2 is not greater than that for diet 1 (i.e., μ_2 < or equal to μ_1) *verses* the alternative hypothesis (H_a) that digestibility of phosphorus in diet 2 is greater than that for diet 1 (i.e., $\mu_2 > \mu_1$). In that case the critical table *t* value (2.353) would be found in the 0.05 column of **Table A-24** (Appendix) with 5% per tail with 3 degrees of freedom. Because the observed *t* (4.78) is greater than the critical table *t* (2.353), we reject the null hypothesis and accept the alternative hypothesis ($\mu_2 > \mu_1$) and conclude that the digestibility of phosphorus in diet 2 is significantly greater than that in diet 1 at the 95% confidence level ($P < 0.05$) using a one tailed test.

An Additional Note About the *t*-Tests for Unequal Variances:

In practice, variance as a measure of variability of unknown individual values for any mean can be calculated as long as you compute variance from other common measures of variability often reported in the literature. Therefore it is easy to compare any mean from your own data with any mean (reported in the literature or wherever) as long a they also report the number of observations (n) and a measure of variability that can be used to calculate variance for a *t*-test for unequal variances. Such measures of variability are usually the standard deviation (s) or the standard error of the mean (SEM) from which variance can be computed. For instance, the standard deviation (s) = the square root of variance s^2. Therefore, variance equals the square of standard deviation. Also the standard error of the mean (SEM) = $\sqrt{(s^2/n)}$. Therefore, variance equals the product of n times the square of SEM, i.e., $s^2 = n \times (SEM)^2$.

Reference:

Ruxton, G.E. 2006. The unequal variance *t*-test is an underused alternative to Student's *t*-test and the Mann-Whitney U test. Behavioral Ecology 17(4): 688-690.

Factors to Investigate Prior to Purchasing a Site

1. Determine from local zoning officials if your intended operation is an allowed use. Be aware of the differences in law between zoning restrictions and agricultural use allowances. Consult a knowledgeable expert in this area. Obtain in writing from a local zoning official that your intended operation is an allowable use activity.
2. Identify, review, and become familiar with all necessary permits (US, state, and local) required for your planned operation, e.g., hatchery permit, stocking permits, waste water discharge permits, food processing permits.
3. Determine the quantity and quality of available water to the site (usually means drilling test wells) and submitting a water sample to qualified laboratory for extensive determination of water quality parameters (see Chapter 2). Is there a backup supply of water (quantity and quality and required treatment costs prior to use) in the event of the primary source becoming depleted or lost? What is the cost to bring water to the production site (Is gravity flow possible or will it require pumping? What are the elevation differences?)
4. Determine the technical feasibility of and cost of disposing of both liquid and solid waste being generated on site. Will the site allow land application of solid and liquid waste and for how many months of the year?
5. Determine the local utility costs for electricity (and availability of 3-phase power), heating fuel, water, sewer, and oxygen. (Different political districts may be dramatically different in utility costs even within the same state.) Are other utility services available, e.g., phone, internet access?
6. Determine whether the load capacity of roads and bridges servicing the farm are adequate to handle semi-trucks.
7. How much land is available for purchase? Can you obtain an option on a future purchase if you wish to expand?
8. Does the site allow (zoning) on-site living quarters and housing? How many people or independent families?
9. How much additional infrastructure cost will be involved to make the site usable for your intended operation, e.g., roads, driveways, drainage?
10. What is the previous history of the site and prior use? Were there any toxic chemicals ever used at the site?
11. How compatible is your operation with the surrounding activities? Even if your operation can be approved for legal usage, you will want to be in a friendly environment for long-term success.
12. How close is your farm to an international airport? This is particularly critical if you will ship or receive live animals (fingerlings). Do you have easy access to an express delivery service, e.g., UPS, FEDEX, DHL? Very helpful for receiving repair components or in shipping product from farm.

CALCULATING VOLUME OF TANKS

Volume measurements are required to determine the proper concentration of chemicals and to calculate holding and transport densities.

Rectangular Tank Volume:

Volume = length · width · depth or L · w · h

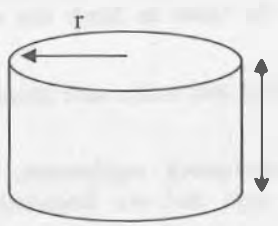
When measuring a tank, take inside measurements of length and with and the depth at the appropriate water level. If a standpipe or other overflow drain is present, then the height of the standpipe is the correct depth measurement. If the bottom of the tank is sloped toward the drain, an average depth measurement should be determined. To find the average depth, take measurements at the two ends and in the middle, add together and divide by 3.

To convert from cubic feet or cubic inches, the conversion table included in the appendix can be used.

Circular Tank Volume:
Circular tank volume is determined by the formula:

Volume = 3.14 · (radius)^2 · depth or $\pi r^2 \cdot h$

The radius is measured as ½ of the inside diameter of the tank, at its base. The radius is squared or multiplied by itself.



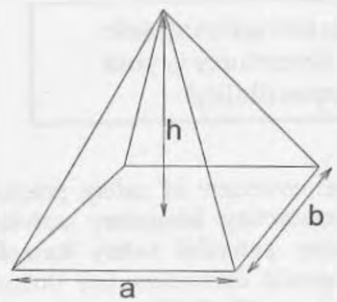
height or depth (h)

Useful conversions:

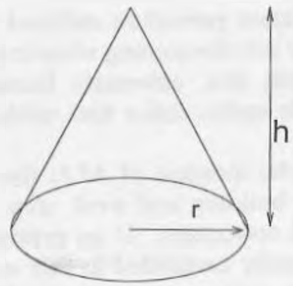
1 gal = 7.481 ft^3	1 ft^3 = 0.1337 gal
1 m^3 = 264.2 gal	1 gal = 0.0038 m^3
1 ft^3 = 1,728 in^3	

Miscellaneous Tank Volume:

Pyramid: Volume = ⅓ * length * width * height or = ⅓* a * b * h



Cone: Volume= ⅓* 3.14 * radius^2 * height or =⅓ * π *r^2 *h



Sphere Segment: Volume= ⅓* 3.14 * height^2 * (3* radius – height) or ⅓ * π *h^2 *(3r-h)

LABORATORY SAFETY PROCEDURES

(Adapted from MCRA Operators Manual, Freshwater Institute, 2000)

> Knowing and utilizing safe research procedures is your responsibility!

This section provides an overview of safety practices that one is expected to follow when conducting laboratory activities. Providing specific information on every potential safety hazard associated is impossible; however, you should customize this document to reflect precautions necessary for your specific facility.

EMERGENCY EVACUATION PROCEDURE

The emergency evacuation procedure outlined in the following paragraphs is applicable to any life-threatening situation you may face at your workplace. For example, fire, extremely hazardous laboratory chemical spills, major pesticide spills, major fuel spills, or propane gas leaks.

It is critical to know the location of **ALL** fire alarms and fire extinguishers throughout your building and work area. When a fire is discovered, be sure to alert all coworkers. If an extinguisher is readily available and the fire can be easily controlled in this manner, do so! If not, pull the nearest fire alarm and evacuate the building through the nearest exit. All employees should exit, meet at a predetermined location, and report immediately to his/her supervisor. **DO NOT** reenter the building until directed to do so by the Station Safety Officer, Director, or maintenance staff.

If time permits after the alarm is sounded, secure any hazardous materials or research procedures you may be involved with, turn off all heat or open flames, and close all doors. If you are disabled, be sure to inform your supervisor so that assistance can be provided upon time of evacuation.

EMERGENCY TELEPHONE NUMBERS

This is an abbreviated list. You should also have numbers for the facility manager, electric company, gas supplier, and other important contacts posted in central locations.

Local Fire, Ambulance, Police	**9-1-1**
National Poison Control Center	**1-800-222-1222**
Chemtrec (chemical spills)	**1-800-424-9300**

ELECTRICAL POWER FAILURE (BLACK OUT)

Fume hoods are an important part of the fresh air circulation systems in laboratory buildings. During a black out, fresh air supply is interrupted, foul air remains in the building, and health hazards may persist. If a black out occurs while you are working with a toxic chemical under the fume hood, cap the container, close the hood sash, and contact your supervisor or director of the laboratory.

EMERGENCY RESPONSE EQUIPMENT

- Know fire alarm and fire extinguisher locations
- Know how to use an extinguisher
- Know which extinguishers are made for which types of fires
- Know where emergency showers and eye wash stations are available—any water source may be used to flush the eyes, i.e., drinking fountains, water taps, etc.
- Know where First-Aid kits are located and make sure items are kept up-to-date
- Never be embarrassed to use emergency equipment; they are provided to keep each employee safe and are intended to keep serious problems to a minimum

BASIC RULES FOR LABORATORY SAFETY

- Keep laboratory aisles and doorways free of boxes, carts, equipment, etc.

- Keep cabinet doors closed and equipment away from edge of lab benches
- Do not store glassware, metal instruments, or heavy items on overhead shelves or on top of cabinets
- **NEVER** place broken glass, sharps, or syringe needles in the household laboratory trash receptacles. Make sure these items are placed in appropriately labeled receptacles
- **NEVER** mouth-pipette any chemical or any substance, including water; ALWAYS use mechanical suction devices
- **ALWAYS** wear proper protective clothing when conducting research (closed shoes, gloves, lab coats, safety goggles, face, shields, respirators, etc.)
- Label all prepared solution containers with contents, date of preparation, and any hazard warnings
- **STUDENTS** are to be under direct supervision when working after normal working hours and on weekends
- Smoking is not permitted in any research facility; smoke only in designated areas outside of the building
- **NEVER** store, prepare, or consume any food or drink in any laboratory where hazardous materials are being utilized

LABORATORY CHEMICAL SAFETY

USE AND HANDLING

It is the responsibility of employees and cooperators to know hazards, properties, and precautions for safe handling of chemicals they are using. ALWAYS READ THE LABELS before using any type of chemical. MATERIAL SAFETY DATA SHEETS (MSDS) should be consulted to find complete information on a chemical or pesticide's properties, hazards, and safe handling methods. Hardcopy MSDS information should be available at the entrance of any laboratory.

Storage of Chemicals

All containers holding prepared solutions or materials should be labeled with a hazard warning if applicable, contents, and preparation date. Large quantities (>l gal) of flammable solvents or concentrated acids/bases should not be stored in your lab; keep only enough for your daily use requirements. Any flammable liquids requiring refrigeration should be done so only in specifically approved refrigerators/freezers. Storage cabinets are provided for the compatible storage of all bulk chemicals and are labeled either by contents (acids, bases) or by scientist's name (solvents). Glass bottles of solvents should never be stored near paper, books or boxes, or above head level or on the floor where they are most susceptible to breakage. Store compatible chemicals (solvents, acids, and bases) together using secondary chemical-resistant spill containment, but separate them into individual spill containment areas.

NEVER STORE GLACIAL AND NITRIC ACIDS TOGETHER!

Disposal of Waste Chemicals - Liquid Wastes

Never dispose of any chemical solution; containing even traces of a toxic chemical, by flushing down the sink drain.

ALL HAZARDOUS LIQUID CHEMICAL WASTE MUST BE CAPTURED AND PROPERLY STORED IN LABELED CONTAINERS.

- Plastic-coated, shatterproof glass bottles or DOT-approved polyethylene drums can be obtained from the safety officer to serve as collection containers for hazardous liquid waste.

- All hazardous waste containers are to be stored in an identified area of your laboratory marked "satellite accumulation area."

Disposal of Waste Chemicals - Solid Wastes

HAZARDOUS MATERIALS SHOULD NEVER BE PLACED IN THE NORMAL LABORATORY TRASH!

- Empty chemical bottles and some pesticide containers must be washed clean before disposal.
- All contaminated gloves, disposable clothing, syringes, pipettes, paper goods, gels, etc. must be discarded as a hazardous waste.
- Package all contaminated materials in labeled containers and dispose of by appropriate means.

Laboratory Chemical Spills

All laboratories should have spill neutralizing and spill control materials readily available to respond to "incidental spills"—releases of hazardous substances where the substance can be absorbed, neutralized, or otherwise controlled at the time of release by employees in the immediate release area, or by maintenance personnel. These materials should be disposed of properly through a waste disposal program.

Personal Protective Equipment

Many operations conducted in the laboratory are dangerous. The hazard potential from explosions, implosions, ruptures; liquid splash or generation of toxic vapors should always be given special consideration. Carefully select appropriate eyewear, gloves, respirators, clothing, and safety shield. Always dress sensibly—avoid open-toed shoes, long, loose hair, and loose-fitting clothes.

Eye And Face Protection

- Wear protective equipment to prevent the hazard of flying objects, liquid splash, dust generation, injurious radiation, etc.

- Select equipment that has been designed and tested in accordance with the American National Standard for Occupational and Educational Eye and Face Protection.
- Wear flexible-fitting goggles with hooded ventilation or safety glasses with side shields and a full-face shield to protect against chemical splash and/or flying glass.
- Never wear contact lenses while handling chemicals. This increases the potential for eye injuries by increasing the concentration of chemicals in contact with the cornea in the case of a chemical splash, laminar flow air can cause dehydration of the eye and cause corneal abrasions, and water-soluble chemical vapors can be absorbed by soft lenses creating exposure to the eye.

Emergency Eye Treatment Program

- Know where all eye wash stations are located; treat an eye injury within 15 seconds of occurrence.
- When washing eyes, get under eyelid and roll eyeballs around in order to wash the entire surface.
- **NEVER** use neutralizing solutions including boric acid and out-of-date eye wash solutions for a chemical splash treatment.
- Apply clean, cool, wet, soft pads to eyes after washing until follow-up help is obtained.
- Do not attempt to remove embedded objects; seek assistance from trained medical personnel.
- See Administrative Officer to fill out an Accident Report.
- Flush eye lavages weekly to assure the water is clear, and then report any problems to maintenance staff.

Hand Protection

- Wear gloves to protect against chemical absorption into skin, highly toxic chemicals, and physical hazards such as heat or sharp objects.
- Use gloves designed for chemical protection made from various materials such as rubber, neoprene, nitrile, and PVC.

- Do not use asbestos gloves; select heat-resistant gloves made from ceramic (alumina silica) fiber.
- Use disposable gloves for handling toxic materials, especially carcinogens and tetragens.
- Removal of gloves should be done so as to eliminate any chance of contamination—using fingers of one gloved hand, pinch the material of the other glove at the base of the palm and peel off glove; continue to hold glove; with the ungloved hand reach under the other glove on the palm side of tbe wrist, pinch and peel off the other glove.
- Thoroughly rinse reusable gloves after use.
- Discard torn or punctured gloves.

Respiratory Protection

- Wear respiratory protection whenever toxic chemicals are handled in areas without adequate ventilation.
- Consider the toxicity and concentration of airborne contaminants, the contaminant's warning properties (odor, taste, and irritation), and proper face piece fit when selecting respiratory equipment.
- DO NOT use an air-purifying respirator if airborne contaminants do not have good warning properties.
- Wear full-face piece respirators when airborne contaminants cause eye irritation.
- Select appropriate cartridge/filter.
- NEVER leave respirators uncovered in work area; cartridges/filters and inner surface of face piece may become saturated with contaminants.
- Inspect the exhalation valve system and headbands of respirators and replace any damaged parts.
- Remove cartridges/filters before cleaning.
- Wash face pieces with a warm aqueous solution of a germicidal detergent; rinse in clean, warm water and air dry.
- Store respirators in plastic bags and keep in station respirator storage cabinet or in a clean, dry, cool area.

Information Resources For Aquaculture
Regional Aquaculture Centers

Northeastern Regional Aquaculture Center
Dr. Reginal Harrell, Director
Phone: 301-405-6511
Email: rharrell@umd.edu
University of Maryland
2113 Animal Sciences Building
College Park, MD 20742-2317
Phone: (301) 405-6085;
Fax. (301) 314-9412
Email: nrac@umd.edu
Website: http://www.nrac.umd.edu
Represents: Connecticut, Delaware, Maine, Maryland, Massachusetts, New Hampshire, New Jersey, New York, Pennsylvania, Rhode Island, Vermont, West Virginia, and the District of Columbia

North Central Regional Aquaculture Center (NCRAC)
Dr. Ted Batterson, Director
Michigan State University
13 Natural Resources Building
East Lansing, MI 48824-1222
Phone: 517-353-1962; Fax: 517-353-7181
Email: batters2@msu.edu
Website: http://www.ncrac.org/
Represents: Illinois, Indiana, Iowa, Kansas, Michigan, Missouri, Minnesota, Nebraska, North Dakota, Ohio, South Dakota, Wisconsin

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Represents: Alabama, Arkansas, Florida, Georgia, Kentucky, Louisiana, Oklahoma, Mississippi, North Carolina, Puerto Rico, South Carolina, Tennessee, Texas, Virginia, Virgin Islands

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E-mail: grahamy@u.washington.edu
Website: http://www.fish.washington.edu/wrac
Represents: Alaska, Arizona, California, Colorado, Idaho, Montana, Nevada, New Mexico, Oregon, Utah, Washington, Wyoming

Other Government Resource Centers

National Sea Grant Depository
Pell Library Bldg., Univ. of Rhode Island, Narragansett Bay Campus, Narragansett, RI 02882-1197
TEL: 401/874-6114, FAX: 401/874-6160
Lending library of all publications funded by Sea Grant.

USDA/CSREES/PAS
Dr. Merle Broussard, Director, Dr. Gary Jensen,
National Program Leader, Maxwell Mayeau, Program Specialist.
United States Department of Agriculture
Cooperative State Research, Education, and Extension Service
1400 Independence Avenue SW, Stop 2201
Washington, DC 20250-2201TEL: 202/401-6438, FAX: 202/401-4888
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U.S. Fish & Wildlife Service
1 Arlington Square, Rm. 820, Arlington, Virginia 22203
TEL: 703/358-1715, FAX: 703/358-2210

Alternative Farming Systems Information Center (AFSIC)
http://www.nal.usda.gov/afsic/
The AFSIC is one of 10 information centers at the National Agricultural Library. AFSIC serves as a national clearinghouse for aquaculture information and provides materials for aquafarmers, consumers, industry personnel, educators, government agencies, associations, libraries, the media, students, scientists, and prospective farmers.

Organizations

American Fisheries Society
5410 Grosvenor Ln., Suite 110, Bethesda, MD 20814-2199
TEL: 301/897-8616, FAX: 301/897-8096
http://web.fisheries.org/main/

American Tilapia Association
111 West Washington St., Charles Town, WV 25414-1529
TEL: 304/728-2167 FAX: 304/728-2196
http://ag.arizona.edu/azaqua/ata.html

Aquacultural Engineering Society
c/o USDA/ARS SMAS
Dr. Tim Pfeiffer
5600 US Highway 1 North
Fort Pierce, FL 34946
772.465.2400 x360 Voice
77.466.6590 Fax
http://www.aesweb.org/

Aquaculture Assoc. of Canada
16 Lobster Lane, St. Andrews, NB E3B 3T6 Canada
TEL: 506/529-4766, FAX: 506/529-4609
http://www.aquacultureassociation.ca/

Catfish Farmers of America
1100 Highway 82 East, Suite 202, Indianola, MS 38751
TEL: 662/887-2699, FAX: 622/887-6857
http://www.catfishfarmersamerica.org/

European Aquaculture Society
Slijkensesteenweg 4, B-8400 Oostende ,Belgium
Tel. +32 59 32 38 59, Fax +32 59 32 10 05
http://www.easonline.org/about/en/contactus.asp

National Aquaculture Association
111 West Washington St., Suite 1, Charles Town, WV 25414-1529
TEL: 304/728-2167, FAX: 304/728-2196
http://www.nationalaquaculture.org/

US Trout Farmers Assoc.
111 West Washington St., Charles Town, WV 25414-1529
TEL: 304/728-2189, FAX: 304/728-2196
http://www.ustfa.org/

US Aquaculture Suppliers Assoc.
PO Box 24866, Little Rock, AR 72221
TEL: 909/652-2612, FAX: 909/652-2492

The U.S. Marine Shrimp Farming Program
Oceanic Institute
41-202 Kalanianaole Hwy
Waimanalo, HI 96795
http://www.usmsfp.org/

World Aquaculture Society
143 JM Parker Coliseum, LSU, Baton Rouge, LA 70806
TEL: 504/388-3137, FAX: 504/388-3493
http://www.was.org/main

DIRECTORIES

Aquaculture Magazine Buyer's Guide,
PO Box 2329, Asheville, NC 28802
TEL: 704/254-7334, FAX: 704/253-0677, $16.00.

Canadian Aquaculture Buyers Guide
Harrison House Publishers, 4623 William Head Rd., Victoria, British Columbia, V9C 3Y7, CANADA
TEL: 250/478-9209, FAX: 250/478-1184, $7.00.

WORLD WIDE WEB

Annual Recirculating Aquaculture Systems Short Course
http://www.bee.cornell.edu/outreach/aquaculture/shortcourse.cfm

American Tilapia Association
http://www.tilapia.org
Access to information about the fish, which is the fastest growing aquaculture crop in the United States and around the world. We are pleased to provide this information to those already producing tilapia for the food industry, for those interested in joining the industry and to potential customers and consumers of farm-raised tilapia.

Aquaculture without Frontiers
http://www.aquaculturewithoutfrontiers.org/
Aquaculture without Frontiers (AwF) is an independent non-profit organization that promotes and supports responsible and sustainable aquaculture and the alleviation of poverty by improving livelihoods in developing countries. Formed in 2004, AwF is registered as a charity in the UK and as a non-profit organization in the USA. AwF has been established for the specific purpose of promoting and supporting responsible and sustainable aquaculture to assist in poverty alleviation through improving rural livelihoods in developing and transition countries. In its work, AwF draws on the experience of respected professionals from every relevant discipline. AwF already has a database of more than 80 volunteers.

Aquacultural Engineering Society
http://www.aesweb.org/

AquaNIC - Aquaculture Network Information Center
http://aquanic.org/
An excellent clearinghouse of aquaculture information available on the internet. It also provides information on how to obtain aquaculture information that is not on the internet.

Fish-Vet
http://users.jaguNET.com/~fishvet/
Information on Fish-Vet (tm), a multimedia program for fast and accurate determination of abnormal conditions in fish keeping, including disease, environmental, and nutritional problems, and the veterinary consultancy service of Fish-Vet, Inc.

National Fisheries Institute

http://www.nfi.org

National Fisheries Institute is the largest trade association serving the US seafood industry. NFI has always been a clearinghouse of business-related information. This website offers dozens of links to government statistics, seafood companies and other web resources. A members-only section adds promotional materials, HACCP information and the latest news on import alerts, legislative updates and more.

Natural Resource, Agricultural, and Engineering Service (NRAES)

http://www.nraes.org

Sea Grant, DOC/NOAA

http://www.seagrant.noaa.gov/themesnpa/aquaculture.html

Sea Grant research and outreach efforts focusing on systems development, genetics, physiology, endocrinology, among many others, have contributed to the creation of several new aquaculture-based industries. These industries include the Gulf of Mexico and South Atlantic soft shell crab industry, the Pacific Northwest oyster and clam industry, the hybrid striped bass industry, and the Mid-Atlantic hard clam industry. In addition, Sea Grant investments have helped to establish new businesses throughout the United States, and have provided improved technologies to these businesses

Seafood NIC

http://www-seafood.ucdavis.edu/home.htm

Hosted by the Sea Grant Extension Program at UC Davis, the Seafood Network Information Center or Seafood NIC, is an on-line home to the HACCP Alliance. It offers page after page of seafood-safety information, as well as training materials, seminar schedules and the FDA's official fishery hazard and control guides. The site provides generic HACCP plans for scores of seafood products and processes.

U.S. Dept. of Agriculture, Agricultural Research Service

http://www.ars.usda.gov/research/programs.htm

The mission of the Aquaculture Program is to conduct high quality, relevant, basic and applied aquaculture research and technology transfer to create jobs and economic activity that will improve the international competitiveness and sustainability of United States aquaculture, and reduce dependence on imported seafood and threatened ocean fisheries.

U.S. Trout Farmers Association

http://www.ustfa.org/

Trout information for both the consumer and industry. Information for the consumer includes: Tips and handling, farm-raised trout, trout tips, handling how-to's, prep pointers, and nutritional values. Industry information includes: Salmonid Magazine, Quality Assurance, membership info, and other internet resources, as well as a trout recipe book.

World Aquaculture Society (WAS)

http://www.was.org/

An international nonprofit society with over 4,000 members in 94 countries. Founded in 1970, its primary focus is to improve communication and information exchange within the diverse global aquaculture community.

SOFTWARE PROGRAMS

The software programs listed and briefly described below are available at: www.bee.cornell.edu/aqua.

Spreadsheets
- Oxygen Design 5
- Fish Tank Design 12
- Pipe Flow Design 19
- Cost Analysis 25

DOS Based
- CO_2 Control Options 31
- LHO Design & Management 54
- AIRPUMP 64

There have been a lot of equations presented in our text Recirculating Aquaculture. By now, you should have a firm grasp on the principles involved in and the logic behind the equations. Someone (probably an engineer!) once said that aquaculture is mostly doing calculations. Well, to make your life easier, we have a collection of spreadsheets and DOS programs that can take a lot of the drudgery out of the calculation steps.

The software programs are available online at www.bee.cornell.edu/aqua

The spreadsheet programs (referred to as workbooks) are as follows:

- Oxygen Design: Calculates all parameters for water flowing from a biofilter, through a spray tower, and back into the tanks. These values include oxygen demands, inflow and outflow concentrations of oxygen and nitrogen, and other water quality parameters.
- Fish Tank Design: Calculates values needed for the design of the tank and the biofilter, including flow rates, and the areas and volumes required for different production rates. There is also a worksheet in the Fish Tank Design workbook that calculates feeding rates and associated growth rates based upon water temperature. This worksheet can be used to forecast growth rates that can be expected, and serve as a basis of comparison of expected versus actual growth rates in the fish tank program.
- Pipe Flow Design: Calculates values relative to the design of the pipes, including drainage pipes and flow volumes. Calculations include velocities, areas, and flow rates for different pipe sizes and the corresponding head pressure requirements.
- Cost Analysis: Calculates, based on specifications given by the user, the fixed and variable costs of facility operation and costs per pound of fish produced for the specified aquaculture facility.
- pH-Alkalinity-CO_2-Temperature: Calculates CO_2 given the other parameters. There is a freshwater and a salinity dependent versions. Do NOT use the salinity dependent version to extrapolate to freshwater conditions (probably reasonably accurate down to ~2 ppt).
- Overall System Design, Staging, and Controlling Flow Rates: A very powerful spreadsheet that does all the growth calculations as affected by temperature and then calculates sizes of tanks necessary to accommodate a series of stages to go from initial stocking to harvest sized animals. Additional worksheets then calculate the required flow rates based upon the constraints specified for oxygen, TAN, CO_2, and TSS. This spreadsheet is often being revised by the authors.

The DOS based programs are:

- CO_2 Control Options: An extensive program that analyzes several control options and associated costs for removing CO_2, plus the additional cost of restoring heat to the building that is lost during ventilation of the removed CO_2.
- LHO Design & Management: **L**ow **H**ead **O**xygenators can be designed with this quick-basic software. Calculates operating efficiencies, total gas pressures, and quantity and

cost of oxygen transferred. This program can be used to optimize design of LHO's or to predict consequences of changing operating parameters.

- AIRPUMP: This program can be used to design airlift and aeration systems, including the proper pipe dimensions and airflow rates for achieving the desired oxygen transfer and water flow rates.

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DEFINITION OF TERMS USED IN AQUACULTURE

Absolute growth - This is growth measured in grams/day.
Acclimation - The process of metabolic compensation in response to change in an environmental factor.
Aerobic - This refers to chemical or biological processes which take place in the presence of oxygen.
Allele - One of two or more forms of a given gene.
Alkalinity – A measure of the buffering capacity of the water.
Ammonia - NH_3. Sometimes this term is loosely used to refer to both forms of total ammonia present in the water (NH_3 and NH_4^+).
Ammonia volatization - This is the loss of ammonia (NH_3) directly into the atmosphere resulting from aeration (called Air stripping). Ammonia is volatilized more readily at high pH values.
Ammonia-nitrogen - This is the amount of ammonia expressed in terms of nitrogen concentration. The molecular weight of ammonia (NH_3) is 17 and the molecular weight of nitrogen is 14. Therefore, 1 mg/L of ammonia-nitrogen = 1 x 17/14 or 1.2 mg/L of ammonia.
Ammonification - This is an aerobic or anaerobic heterotrophic or autotrophic process that deaminates proteins and produces ammonia-nitrogen.
Ammonium - NH_4^+. This is the predominant form of the ammonia equilibrium.
Anaerobic - A chemical or biological process that takes place in the absence of oxygen.
Aquaculture - The controlled production of a crop in water.
Autosome - A chromosome other than the sex chromosome.
Backcross - Crossing an Fl hybrid heterozygote back to one of the parental types.
BOD_u – A measure of the total amount of oxygen required to stabilize a waste biologically, ultimate carbonaceous biochemical oxygen demand.
Button-up - Fingerling closes yolk sac.
Broodfish - Fish used to produce fish that will be cultured.
Carrying capacity -Biomass in a given area where growth stops (lbs/ft^3, kg/m^3).
$cBOC_5$ – A measure of the amount of oxygen required to stabilize a waste biologically, five-day carbonaceous biochemical demand.

Clarification - The process of removing suspended solids from water.
COD – Chemical oxygen demand, often used as a substitute for BOD tests.
Cohort - A group of fish of the same age or size stocked in the system at the same time.
Critical standing crop - The inflection point on a growth curve indicating the presence of a growth limiting factor.
Denitrification - This is the anaerobic process by which chemotropic bacteria convert nitrates (NO_3^-) to nitrogen gas (N_2), N_2O or ammonia (NH_3).
Digestible Carbohydrate- A value that indicates the utilizable amount of carbohydrates. For salmonids the following are digestibility values for different types of carbohydrates: simple sugars 100%; complex sugars 90%, cooked starch 60%; raw starch 30%; fiber 0%. (Piper et al. 1989, 219)
FDA - United States Food and Drug Administration
Feed conversion ratio - This is the ratio of the amount of feed fed (moisture content < ~ 5%) to the amount of weight gained for a group of fish (moisture content of a whole fish is ~ 80%).
Flashing - Fish rolling over upside-down and exposing their ventral side. Fish swimming at the surface appearing to gasp for air.
Flow Through Rate - The volume of new water per unit time passing through a culture tank; refers to the make-up water specifically.
Fry – the fish shortly after hatching (fry become fingerlings)
Genotype - Genetic determination of sex.
Grading - The process of sorting fish by size.
Gross yield - Total weight of fish harvested for a volume of water.
Gynogenesis - Producing all female fish.
Hapa - A net enclosure used to hold fish that allows water circulation. It is usually suspended in a tank or pond.
Harvest - The removal of harvestable sized fish or the removal of fish from any stage of the production cycle.
Haploid - Half the normal number of chromosomes.
Heterogametic - Fish whose sex determining alleles are different, i.e., XY or WY.
Homogametic - Fish whose sex determining alleles are the same, i.e., XX or WW.
Hybrid vigor - Exceptional growth ability of the first generation (F1) resulting from a cross of two different species.
Low Regulatory Priority (LRP) - Drugs with this status are not officially approved by the FDA. The FDA is "unlikely to object" to these compounds if used under the exact conditions specified. The FDA has not made any statements regarding the safety or efficacy of these products for the purpose under which they are Low Regulatory Priority.
Mean Hydraulic Residence Time (HRT) - Refers to the time required at a given rate of flow for a complete volume of water in a tank to be exchanged, V (volume of tank) / Q(flow rate).
Meiosis - The process by which chromosomes replicate, form homologous pairs, and then segregate into different nuclei to produce haploid cells.

Mortality rate - This is the calculation of the number of fish that die versus the number of fish stocked for any part (or all) of the production cycle.
Mitosis - A process of cell division in which the chromosomes replicate and divide equally so that identical daughter cells are produced.
Net yield - Total weight harvested minus total weight stocked for a volume of water (lbs/ft^3, kg/m^3).
Necropsy - A postmortem internal and external examination of fish.
Nitrate-nitrogen - This is the nitrite concentration expressed in terms of its equivalent nitrogen concentration. The molecular weight of nitrite is 62 and the molecular weight of nitrogen is 14. Therefore, 1 mg/L of nitrite-nitrogen = 1 x (62/14) or 4.4 mg/L of nitrate.
Nitrification - The aerobic process by which bacteria convert ammonium (NH_4^+) to nitrates (NO_3^-). Nitrification is most rapid at pH of 7–8 and at temperatures of 25–30°C. Nitrification causes waters to decrease in pH.
Nitrobacter - The nitrifying bacteria responsible for converting nitrites (NO_2^-) to nitrates (NO_3^-).
Nitrosomonas - The nitrifying bacteria responsible for converting ammonium (NH_4^+) to nitrites (NO_2^-).
Osmotic regulation - This is a process by which freshwater fish are able to excrete excess water yet retain ions and substances vital to normal function. There is a constant flow of water into their bodies caused by the difference in ion concentrations between the inside and outside of their body. Saltwater fish have the opposite problem. They are continually losing water from their bodies and must work to retain water needed for normal function.
Phenotype - Secondary characteristics (not genetic makcup) that indicate sex, i.e., genitals, size, color.
Polishing filter - These are secondary filters that remove fine solid materials from water that has already passed through a primary filter (ozone, ultraviolet filters, cartridge filter, drum filter, foam fractionator).
Polyploidy - Other than the normal number of chromosomes, i.e., triploids, tetraploids.
Production to Capacity Ratio (P/C ratio) - This is the ratio of system output per year to the maximum carrying capacity of the system. P/C ratios of 3 are typical of high density systems.
Purging - A process that removes any "off flavor" in harvestable sized fish by holding fish unfed in clean water for a few days.
RAS – Recycle (Recirculating Aquaculture Systems) See Recycle below.
Recycle Percentage - The percentage of the total system volume that is retained, on a daily basis.
Reuse (serial reuse) - Water is reused in multiple tanks, moving in one direction never used in the same tank twice (as NOT recycled); often referred to as serial reuse.
Recycle (Recirculating Aquaculture Systems or RAS) - Water flows from a tank (s) to a treatment process and then is returned to the tank, hence the term recirculated or recirculating aquaculture systems or RAS. RAS are

generally regarded as systems that discharge less than 20-50% of the standing water in the system volume per day. Some RAS may discharge more water, but become increasingly more like a reuse system.

Relative fecundity - This is a measure of egg production from broodfish. It is the number of eggs received per spawn for each kilogram of broodfish weight.

Relative growth - Growth measured as a percent of the fishes body weight.

Replacement Percentage - The percentage of the total system volume replaced per day.

Sac Fry - Early stage fingerlings, still on egg sac.

Serial Reuse– See Reuse.

Settleable solids - This is the volume or weight of solid material that will settle in one hour from one liter of water confined in a cone shaped cylinder called an Imhoff cone, ml/L.

Sex chromosome - A chromosome, usually X or Y that plays a major role in the determination of sex.

Species - A population or populations of individuals that share a common gene pool, are phenotypically similar, and are reproductively isolated from other species.

Specific Surface Area - Surface area of the media per unit volume; usually referring to the surface area of a particular media used in filtration or settling components

Standing crop - Biomass in a given volume of water at a particular point in time (lbs/ft3, kg/m3).

Stress - A physiological response caused by an external stimulus (stressor) that results in an energetic cost to the organism.

Supermale - A male carrying only male determining genetic information (YY).

Survival rate - The calculation of the number of fish that remains the number of fish stocked for any part or all of the production cycle.

System - A system refers to a complete aquaculture production unit that includes: production tanks, filtration and circulation.

Stocking Density - Mass of cultured product per volume of tank (ignores the effects of fish displacing part of the water volume)

Tetraploids - Double the normal chromosome number.

Total Biomass - Mass of cultured product in the culture system.

Total dissolved solids - This is the total residue (mg/L) left after evaporation of a filtered water sample.

Total System Volume - Volume of water in the culture tank, pipes, reservoirs, treatment tanks, and pumps.

Toxic ammonia - This refers to ammonia (NH_3), which is lethal to fish at fairly low concentrations.

Total Solids (TS) – The residue remaining after sample has been evaporated and dried at a specified temperature (103 to 105 °C)

Total volatile solids (TVS) – Solids that are volatilized and burned off when the TS are ignited (500±50 °C).

Total suspended solids (TSS) – Portion of the TS retained on a filter (most often a Whatman glass fiber filter) with a specified pore size (1.58 μm) after drying at 105°C.

Triploid - 1.5 the normal number of chromosomes.

Volatile suspended solids (VSS) – Solids that are volatilized and burned off when the TSS is ignited (500±50 °C).

SUMMARY LIST OF SYMBOLS

a_{TSS}	TSS produced as a proportion of feed fed (kg TSS per kg feed)A Cross-sectional area or area, m^2 (ft^2)
A	Total hole area over the entire LHO, m^2
Ao	Constant used for regression in water vapor equation, dimensionless
A_{sz}	Settling basin floor area (settling zone only), m^2
BHP	Brake horsepower, kW (hp)
BL	Fish body length, dimensionless
BOD_5	Biological Oxygen Demand 5 day, mg/L
c_p	Specific heat of air, kJ/kg°C (BTU/lb°F)
C	Concentration of solids in tank water, kg/m^3
C	Coefficient relating pressure to velocity, dimensionless
$C_{density}$	1.5 for L in inches (0.24 for L in cm) for tilapia $C_{out,b}$ concentration of pellets leaving bottom center drain, kg/m^3
C_D	Drag coefficient, dimensionless
C_d	Discharge coefficient to predict velocity through an orifice, dimensionless
CF	Condition factor that relates fish weight and length, dimensionless
$C_{H.W}$	Roughness constant used in Hazen-Williams
C_i	Concentration of gas species "i", mg/L
C_{in}	Influent dissolved concentration of a gas species, mg/L
$C_{meas,i}$	Measured concentration of gas "i" in water, mg/L
COC	Cumulative oxygen consumption, kg
COD	Chemical Oxygen Demand, mg/L
C_{out}	Effluent dissolved concentration of a gas species, mg/L
$C_{s,i}$	Dissolved gas saturation concentration for species "i", mg/L
C_x	Concentration of a particular water quality parameter

X, $\frac{kg\ of\ X}{10^6\ kg\ water}$

C_0, C_1, and C_2 Concentrations of parameter X crossing the control volume, mg/L

date Julian day of the year as a number
D cumulative depth of wastewater applied, m
D Diameter of pipe, ft (m)
D Depth of sedimentation basin, m
$D_{density}$ Carrying capacity or biomass density, kg/m^3 (lb/ft^3)
DDR Diameter depth ratio of tank, dimensionless
D_{fish} Fish density in culture vessel, kg/m^3
D_{inch} Diameter of pipe, inch
DN Dissolved nitrogen concentration, mg/L
DO Dissolved oxygen concentration, mg/L
D_p Diameter of particles, m
D_1 Diameter of upstream section, cm (in)
D_2 Diameter of throat or downstream section, cm (in)

E Gas transfer efficiency, %
E_{pump} Pump efficiency, %
E_{TKN} Enrichment factors for TKN
E_{TSS} Enrichment factors for TSS
E_{VS} Enrichment factors for volatile solids

f Friction factor (a function of the Reynolds number)
f_{rem} fraction of solids removed
f_T Frictional loss coefficient that depends upon the size of the fitting
F Gas mean residence time for one flushing to occur for a chamber, hr
F Perimeter heat loss term, W/K·m (BTU/°F·ft·h)
F Settling zone safety factor
F Feeding rate, kg/day
$F_{\%}$ Feeding rate per day as a percentage of fish body mass, %
FG Feed to gain ratio, dimensionless
$FITTING_{constant}$ Frictional loss coefficient that is size independent of the fitting

g Acceleration of gravity, 9.81 m/s^2 (32.2 ft/s^2)
G/L Gas to liquid ratio, both being on a volumetric basis,

G_T Overall mass transfer coefficient at a specific temperature T °C, dimensionless
G_{20} Overall mass transfer coefficient at 20°C, dimensionless

h height above sea level, m
h Correction for channel kinetic energy, m (ft)
H Dynamic heads ($v^2/2g$), m (ft)
HL Hydraulic loading, kg/m^2 per s
H_L Lost energy head due to friction, m (ft)
HRT Hydraulic residence time, 1/h

J_i Constants used to calculate gas partial pressure due to a specific gas "i" dissolved to some concentration in water, dimensionless

k 1st order rate constant characterizing bead enrichment at the bottom-center drain
K Resistance coefficient for particular style of fitting
K_i Ratio of the molecular weight to volume (mg/mL)
KS saturated hydraulic conductivity, cm/d

L Loading rate, kg/m^3/hr
L depth of manure layer or material providing the resistance to flow, m
L Length of pipe, m (ft)
L Fish length, inches (cm)
L_{oxygen} Allowable fish biomass loading due to oxygen availability, kg/ Lpm
$L_{raceway}$ Length of the raceway in meters

$\dot{m}$ Airflow rate, lb/hr (kg/hr)
MRT_{ideal} Ideal mean residence time, 1/h

N number of applications of wastewater

o.d. Outside diameter of a particle or other object, length unit

P_i^l Partial pressure of gas "i" in liquid form, mm Hg
P_i^g Partial pressure of gas "i" in gas phase form, mm Hg
$p_{wv,inside}$ Vapor pressure inside air, inches mercury water gauge (mm Hg)

$P_{wv,outside}$	Vapor pressure outside air, inches mercury water gauge (mm Hg)
P	Pressure of the fluid, psig (kPa)
P	Perimeter length of exposed walls, m (ft)
P:B	Ratio of yearly production to biomass carrying capacity
P_{BP}	Barometric pressure, mm Hg
PC	Protein concentration or feed, decimal
P_{CO2}	Production rate of carbon dioxide, kg/day
P_{Oxygen}	Production rate (negative as related to feed consumption) of oxygen, kg/day
$P_{Solids, TSS}$	Production rate of total suspended solids, kg/day
P_{TAN}	Production rate of total ammonia nitrogen, kg/day
P_{TSS}	TSS production rate (kg TSS produced per day)
P_{TG}	Total gas pressure, mm Hg
P_{WV}	Water vapor pressure, mm Hg
P_1	Upstream pressure, psi (kPa)
P_2	Throat pressure, psi (kPa)
Q	Inlet flowrate, m^3/s
Q	Water flow, m^3/time
Q	Rate of gas flow, kg/time
Q	Mass flow, lb/s (kg/s)
Q	Heat loss or gain, BTU (kJ)
Q_{air}	Air flow rate through column, m^3/s
$Q_{air,Lpm}$	Air flowrate, Lpm
Q_{heater}	Sensible heat added by space heaters, BTU/h (kJ/h)
Q_0	Flow rate passing through culture tank (discharge), m^3/day (as kg/day)
$Q_{out,h}$	Flowrate flushed through the bottom center drain, m^3/h
Q_{out1}	Flow rate leaving the sidewall drain (m^3 per day)
Q_{out2}	Flow rate leaving the bottom center drain (m^3 per day)
Q_s	Sensible heat production of fish, BTU/h (kJ/h)
Q_{solar}	Solar heat gain, BTU/h (kJ/h)
Q_m	Sensible heat added by motors and lights, BTU/h (kJ/h)
Q_{vi}	Sensible heat ventilated into air space, BTU/h (kJ/h)
Q_{evap}	Rate of sensible heat converted to latent heat via evaporation, BTU/h (kJ/h)
Q_{wall}	Sensible heat conducted from the space through walls and ceiling, BTU/h (kJ/h)
Q_{floor}	Sensible heat lost through the floor, BTU/h (kJ/s)
Q_{vo}	Sensible heat ventilated out of air space, BTU/h (kJ/h)
Q_1	Water that is recirculated, kg/day

r_{feed}	Feeding rate (kg feed per kg fish per day)
R	Gas flow rate into an LHO chamber, L/hr
R	Number of water exchanges per hour through the rearing unit, hr^{-1}
R	Thermal resistance, (hr°F ft^2)/BTU ($m^2\cdot$°K/W)
RH	Relative humidity, %
R_{vs}	Removal rate of volatile solids, g/day
$R_{pressure}$	Resistance to gas transfer
R_{H2O}	Resistance to water vapor flow; inverse of permeance or perms, hr ft^2 inch Hg/grains (g-m/(24 h m^2 mm Hg))
SS	Suspended solids, mg/L
$S_{\%}$	Percent saturation of a particular gas, %
t	Time
t	Time period for feeding daily ration, day
T	Temperature, °C (°F)
T_a	Average temperature, °F (°C)
TAN	Total Ammonia Nitrogen, kg
T_{base}	Temperature base, usually 65°F (18.3°C)
T_{base}	Equivalent to the temperature at which no growth occurs, °F (°C)
T_{db}	Dry bulb temperature, °F (°C)
T_{dp}	Dew point temperature, °F (°C)
TDH	Total dynamic head that pump operates against, ft (m)
T_{eff}	Treatment efficiency (removal or addition), %
TGP	Total gas pressure, mm Hg
TKN	Total-Kjeldahl-Nitrogen, (Org N + NH_4^+)
TN	Total Nitrogen, mg/L
TOC	Total Organic Carbon, mg/L
TP	Total phosphorus, mg/L
TSS	Total suspended solids, mg/L
$TSS_{capture\%}$	Percent removed of incoming TSS, %
TSS_{in}	TSS concentration entering unit (kg/m^3 or ppm)
TSS_{out1}	TSS concentration leaving the sidewall drain (kg/m^3)
TSS_{out2}	TSS concentration leaving the bottom center drain (kg/m^3)
TU_{base}	Temperature units required to produce an increment of growth per unit time, °C (°F)

ΔT — Temperature difference inside air film to outside air film, °C (°F)

TVS — Total volatile solids, mg/L

U_{max} and U_{min} — Maximum and minimum monthly average values for particular weather variable

V — Volume of single mixing chamber in an LHO, L

V — Fluid velocity, m/s (ft/s)

V_{basin} — Basin volume, m^3

V_o — Volumetric flow of water per unit surface area, m^3/m^2 time culture volume)

V_{orif} — Velocity through the water inlet structure (orifices or slots), m/s

$V_{raceway}$ — Raceway velocity, cm/sec

V_{rota} — Rotational velocity in the tank, m/s

V_{safe} — Fish body lengths per second, BL/s

VS — Volatile solids, mg/L

VSS — Volatile suspended solids, mg/L

V_s — Velocity of settling particles, m/s

V_{tank} — Water volume in the tank, m^3 or L

V_1 — Velocity in upstream section, ft/s (cm/s)

V_2 — Velocity in throat of venturi, ft/s (cm/s)

W — Rate of moisture movement, grains/hr (grams/hr)

X_i — Mole fraction of gas (dimensionless)

X_{inside} — Inside air quality parameter, lbs per lb dry air (kg/kg)

$X_{outside}$ — Outside air quality parameter, lbs per lb dry air (kg/kg)

Y — depth of wastewater applied per application event, m

Y_1 — Hydraulic head over flooded plate, cm

Y_2 — Hole diameter, mm

Y_3 — Pool depth, cm

Y_4 — Fall height of water from plate to receiving pool, cm

Z — Packing height of media, m

Z_{tower} — Height of an enclosed spray tower, m

α — Proportionality constant to relate tank and orifice velocity

α — Field water G20 / clean water G20, dimensionless

β — constant used to estimate accumulation rate of manure per unit of waste water applied, m/m

φ_{enrich} — Relative enrichment factor due to Cornell dual drain, %

ρ — Density of water, kg/m3

γ — Specific weight of the fluid, lbs/ft3,

ν — Fluid kinematic viscosity, ft2/s (m2/s) (see Table 2.1)

ρ — Density of the fluid, lb/ft3 (kg/m3) (see Table 2.1)

Ω_{sg} — Specific gravity fluid, 1.00 for water, dimensionless

λ_{solar} — Weather model constant for solar radiation (83)

$\lambda_{temperature}$ — Temperature weather model constant for dry bulb temperature (100)

ϕ — Argument used in weather model based upon Julian date

ABBREVIATIONS/UNITS

ac	acre(s)
ac-ft	acre-feet
A	ampere(s)
ABS	Acrylonitrite Butadiene Styrene
Abs	absorbance
AES	Aeration Efficiency Standard
amp(s)	ampere(s)
ATC	Automatic Temperature Compensation
atm	atmosphere(s)
AWG	American wire gauge
BNC	bayonet electrical connector
BOD	Biological Oxygen Demand
BOD_5	five day Biological **Oxygen Demand**
BOD_u	ultimate Biological Oxygen Demand
BOD_c	carbonaceous Biological Oxygen Demand
BP	barometric pressure
Btu	British thermal unit
cc	cubic centimeter
CD	corona discharge
cfh	cubic foot (feet) per hour
cfm	cubic foot (feet) per minute
cfs	cubic foot (feet) per second
cm	centimeter(s)
COD	chemical oxygen demand
conc.	concentration
CPVC	Chlorinated polyvinyl chloride
cu.ft.	cubic foot (feet)
cu.in.	cubic inch(es)
cu.yd.	cubic yard(s)
d	day(s)
dB	decibel(s)
DI	deionized or deionization
dKH	degree(s) of carbonate hardness
D.O.	dissolved oxygen

dP	differential total dissolved gas pressure	GLP	good laboratory practice
DWV	drain-waste-vest	gn	grain(s)
EC	electrical conductivity	gpd	gallon(s) per day
ETDA	ethylenediamin etetraacetic acid	gpm	gallon(s) per minute
EPDM	ethylene propylene diene monomer	ha	hectare
fc	foot-candles	HDPE	high-density polyethylene
FIPT	female inside pipe thread	HID	high intensity discharge
FLA	full load amperage	hp	horsepower
fl.oz.	fluid ounce(s)	HO	high output
FNPT	female national pipe thread	HPS	high-pressure sodium
fps	foot (feet) per second	hr(s)	hour(s)
FPT	female pipe taper	HUFA	highly unsaturated fatty acids
FRP	fiberglass reinforced plastic	Hz	hertz or cycles per second
ft	foot (feet)	I.D.	inside diameter
ft^2	square foot (feet)	in	inch(es)
ft^3	cubic foot (feet)	in^2	square inch(es)
ft/min	feet per minute	in^3	cubic inch(es)
ft/s	feet per second	I/O	input/output
ft^3/min	cubic feet per minute	ISE	ion selective electrode
ft^3/s	cubic feet per second	J	Joule(s)
g	gram(s)	K	degrees Kelvin
g/m^2	gram(s) per square meter	kcal	kilocalorie(s)
g/m^3	gram(s) per cubic meter (mg/L)	kg	kilogram(s)
gal	gallon(s)	kg/m^3	kilogram(s) per cubic meter
GFIC	ground fault circuit interrupter	KH	carbonate hardness
GH	general hardness	KJ	kilojoule(s)
g/hr	gram(s) per hour	km	kilometer(s)
		kPa	kilopascal(s)
		kW	kilowatts
		kWh	kilowatt-hour(s)
		l or L	liter(s) or length
		lpm	liter(s) per minute
		lps	liter(s) per second
		lb(s)	pound(s)

LDPE	low-density polyethylene	NPSH	net positive suction head
lpm	liter(s) per minute	NPT	national pipe thread
lux	lumen(s) per square meter	NST	national standard thread
m	meter(s)	NTU	nephelometric turbidity unit(s)
m^2	square meter(s)	O_3	ozone
m^3	cubic meter(s)	O.D.	outside diameter
m^3/h	cubic meter(s) per hour	ODP	open drip proof
m^3/s	cubic meter(s) per second	ORP	oxidation /reduction potential
mA	milliampere(s)	oz	ounce(s)
meq	melliequivalent(s)	Pa	Pascal
mg	milligram(s)	PB	Polybutylene
mg/L	milligram(s) per liter	PE	polyethylene
Mgal/d	million gallons per day	pH	potenz (power) H (hydrogen)
MH	metal halide	PP	polypropylene
min	minute(s) or minimum	ppb	part(S) per billion
MJ	megajoule(s)	ppm	part(s) per million
mm	millimeter(s)	ppt	part(s) per thousand
mmHg	millimeter(s) of mercury	psi	pound(s) per square inch
MNPT	mail national pipe thread	pt	pint(s)
mol	mole(s)	PVC	polyvinyl chloride
m/s	meter(s) per second	PVDF	polyvinylidene fluoride
MPT	male pipe taber	qt	quart(s)
N	Newton(s)	RH	relative humidity
N/m^2	Newton(s) per square meter	R/O	reverse osmosis
N.C.	normally closed	rpm	revolution(s) per minute
NEMA	National Electrical Manufactures Association	SAE	standard aeration efficiency
NFT	nutrient film technique	scfh	standard cubic Foot (feet) per hour
nm	nanometer(s)	scfm	standard cubic Foot (feet) per minute
N.O.	normally open	sec	second(s)
		sq.ft.	square foot (feet)
		sq. in.	square inch(es)

SG	specific gravity	TXV	thermostatic expansion valve
SPDT	single pole, double throw	USB	universal serial bus
Spig	spigot	UV	ultraviolet
SS	stainless steel	V	volt(s)
STP	standard temperature and pressure	VA	volt-amp(s)
TAN	total ammonia-nitrogen	VAC	volt(s), alternating current
TDH	total dynamic head	VSC	volt(s), direct current
TDS	total dissolved solids	VOC	volatile organic compound
TEFC	totally enclosed, fan-cooled	W	width or watts
Temp	temperature	Wt	weight
TGP	total dissolved gas pressure	yd(s)	yard(s)
ton	2000 lb mass	°C	degrees Celsius/Centigrade
tsp	teaspoon	°F	degrees Fahrenheit
		"	inch(es)
		µ	micron(s)

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